

STRIVE

Report Series No. 3

The Value of Parasitic Hymenoptera as Indicators of Biological Diversity

STRIVE

Environmental Protection
Agency Programme

2007-2013

Environmental Protection Agency

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The Value of Parasitic Hymenoptera as Indicators of Biological Diversity

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by

Agriculture and Food Science Centre, University College Dublin

Authors:

Annette Anderson and Gordon Purvis

ENVIRONMENTAL PROTECTION AGENCY

An Ghníomhaireacht um Chaomhnú Comhshaoil
PO Box 3000, Johnstown Castle, Co. Wexford, Ireland

Telephone: +353 53 916 0600 Fax: +353 53 916 0699

E-mail: info@epa.ie Website: www.epa.ie

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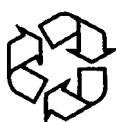
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The EPA STRIVE Programme addresses the need for research in Ireland to inform policymakers and other stakeholders on a range of questions in relation to environmental protection. These reports are intended as contributions to the necessary debate on the protection of the environment.

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Details of Project Partners

Annette Anderson

School of Biology and Environmental Science
Agriculture and Food Science Centre
University College Dublin
Belfield
Dublin 4
Ireland

Tel.: +353 1 7167119

E-mail: annette.anderson@ucd.ie

Gordon Purvis

School of Biology and Environmental Science
Agriculture and Food Science Centre
University College Dublin
Belfield
Dublin 4
Ireland

Tel.: +353 1 7167741

E-mail: gordon.purvis@ucd.ie

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Executive Summary

This study set out to test the hypothesis that parasitoid wasps (Hymenoptera: Parasitica) are suitable bioindicators for, and can provide a useful means to assess, the wider biodiversity of arthropod populations in agro-ecosystems. There is a wide range of theoretical arguments to support such a claim, including the high trophic position of these taxa within the arthropod communities in which they occur, and the unique nature of their biological relationships with the vast majority of terrestrial insect groups.

Currently, grassland can be managed according to Rural Environment Protection Scheme (REPS) specifications by reducing only the intensity of nitrogen use and stocking rates. However, if the widely used method of intensive grass utilisation by short-term rotational grazing and conservation for silage remains unchanged, our data suggest that the net benefit for biodiversity is likely to be small. More detailed studies of alternative grazing and grass utilisation systems are required to maximise the potential biodiversity benefits of the REPS policy.

A total of 178 genera of parasitoid Hymenoptera were identified in agricultural grasslands during the study. These identifications have been verified by independent taxonomic experts, and include ten taxa not previously recorded from Ireland. All taxa, including the new records, were collected from moderately-to-intensively managed grasslands, suggesting that even in such commonplace habitats there may be much Irish biodiversity yet to be discovered. Using multiple data sets, we show that in agricultural grasslands both the abundance and diversity of parasitoid wasp taxa are more closely correlated with overall arthropod diversity than is the incidence of any other insect group, a fact that provides us with a practicable and usable monitoring tool for tracking change in wider arthropod diversity.

Assessment of parasitoid assemblages in existing Teagasc field experiments contrasting different aspects of grassland husbandry (the use of conservation field margins at Johnstown Castle, and the reduction of nutrient inputs and stocking rates at Johnstown Castle and Grange) showed a consistent pattern of management effects on parasitoid abundance, diversity and altered community structure – parasitoid taxa associated with predictable host groups, such as aphids and dung-breeding Diptera, were more prominent in more intensive grassland treatments, while a greater diversity of parasitoids of particularly well-concealed plant-mining and gall-forming insect larval hosts, and insect and spider eggs were characteristic of more extensive management treatments.

The degree to which such management effects are apparent is effectively a reflection of the relative contrast in the ‘intensity’ of the systems being compared. These results support previous work that has shown the overwhelming importance of grassland sward structure on arthropod populations.

The project has confirmed the initial hypothesis and illustrated the bioindicator potential of parasitoid populations. The total abundance of parasitoids is a reliable and relatively easily measured indicator of overall arthropod diversity in agricultural grasslands, providing an eminently user-friendly monitoring tool. However, the project has also shown that a more detailed knowledge of parasitoid taxonomy, biology and host groups can provide a unique ecological insight into underlying environmental influences affecting wider arthropod biodiversity and community structure. With improving knowledge of their host group relationships, parasitoids offer a unique means to study the highly ‘cryptic’ hidden biodiversity that occurs even in the most mundane of habitats, and which would otherwise be almost impossible to assess directly by any other means.

1 General Introduction

1.1 Background

There is currently a great need for, but little agreement on, the selection of suitable biological indicators of terrestrial environmental quality with which to validate agri-environmental policy. This question lies at the core of the Ag-Biota Project (ERTDI 2001-CD/B1-M1), which in addition to addressing a wide range of other issues relating to biodiversity within the agricultural sector, aims to develop appropriate monitoring protocols with which to assess the impact of changing farm practices on biodiversity. The Hymenoptera: Parasitica (parasitoid wasps) represent one of the most numerous and diverse of all insect groups, and have both ecological and economic importance in natural and managed ecosystems (LaSalle, 1993). The great majority of this extremely diverse group of insects are parasitoids of other insect and arthropod species (LaSalle, 1993); the only pterygote orders that are apparently not parasitised are the Ephemeroptera, Plecoptera, Phthiraptera (=Siphonaptera) and Stepsitiera (Askew and Shaw, 1986). Parasitoids differ from conventional parasites in that the completion of their own parasitic development invariably results in the eventual death of their host. Parasitoid wasps that halt the continued development of their host during their own development are called idiobionts. Such species tend to be generalists, parasitising a relatively wide range of host taxa (Quicke, 1997). Other parasitoid species, called koinobionts, have evolved a closer biological relationship with their hosts, involving complex physiological mechanisms that permit continued host development during the development of the parasitoid. This more specialised developmental biology can considerably extend the time available for parasitoid development. Probably as a consequence of their more intimate developmental relationships with their hosts, koinobiont species tend to have a narrower host range than idiobionts (Quicke, 1997). However, as is often the case in biology, there are many exceptions to this generalisation (Askew and Shaw, 1986).

Whether idiobiont or koinobiont, parasitoids in general are a numerous and biodiverse group, with most arthropod groups being hosts to multiple parasitoid species, and many parasitoids being secondary, or even tertiary, hyper-parasitoids (parasitoids of parasitoids) in complex community structures (Sullivan, 1987). Their diversity and specialised ecological relationships with the most diverse of all terrestrial groups (the arthropods) and their significance as natural enemies of insect pests make the parasitoid Hymenoptera a strong candidate bioindicator group, particularly for agro-ecosystems. However, the very diversity that makes this group such a potentially useful tool with which to assess overall biodiversity has, until recently, been a major impediment to this potential. As a consequence of the inaccessibility of taxonomic knowledge of this vast group of insects, ecologists have avoided trying to work with them. However, taxonomic knowledge of most European groups has now progressed to at least the level of genus, but knowledge of individual species is likely to remain inaccessible for some considerable time. However, this is not likely to present a continued obstacle to their meaningful study, since their biology is so specialised that information on their community structure, even at genus level, makes it possible to investigate their potential as bioindicators in an agricultural context.

A preliminary analysis of initial monitoring data collected from the Ag-Biota project, in which parasitoids were only identified to family level, provided support for our original hypothesis that these insects have potential as a means to study effects on overall insect diversity, the statistical correlation between numbers of parasitoid families collected from agricultural grassland, and the taxon richness of all other arthropod taxa being stronger than for any other group. The current study built on this initially encouraging finding, particularly by utilising the increased taxonomic knowledge of European parasitoid groups at genus level within the framework of the original Ag-Biota work programme. In addition to

increasing the taxonomic resolution in Ag-Biota's further exploration of the relationships between parasitoid diversity and wider arthropod diversity, we also test the potential use of existing information regarding parasitoid host ranges as another means to measure, and potentially better understand, the influences of agricultural practice on overall arthropod diversity in grassland.

1.2 Overall Aims

The overall aims of the study were:

- To increase the taxonomic resolution of further monitoring studies on parasitoid Hymenoptera within the Ag-Biota project
- To investigate with the benefit of increased taxonomic resolution, the potential of parasitoid Hymenoptera as bioindicators of other arthropod diversity
- To use the accumulated parasitoid database from ongoing Ag-Biota studies and existing published records of parasitoid host ranges to investigate parasitoid host relationships in monitored sites
- To use the developed expertise in hymenopteran taxonomy and knowledge of host ranges to compare parasitoid assemblages in farm and experimental systems, in order to better understand the ecological influences of agricultural practices on arthropod diversity.

2 Increased Taxonomic Resolution for Parasitoid Hymenoptera Collected in Irish Agricultural Grassland

2.1 Background and Aims

The importance of measuring and monitoring biodiversity was quickly realised following the 1992 Rio de Janeiro Convention on Biological Diversity (CBD) when many authorities began to implement sectoral biodiversity action plans (Magurran, 2004). Initial studies by the Ag-Biota project provided support for the hypothesis that the parasitoid Hymenoptera were a likely candidate bioindicator group for wider insect and arthropod diversity in agricultural grasslands – one of the most widespread habitat types in the Irish countryside. This initial work, however, was limited in that sampled parasitoid populations were identified only to family level. The first objective of the current study was therefore to increase the level of taxonomic resolution in ongoing Ag-Biota studies. Parasitoid Hymenoptera are taxonomically a difficult group to work with. It has been estimated that at least 75% of species have not yet been formally described, and those that have been adequately described to species level require very specialised expertise to identify (LaSalle, 1993). A steadily increasing body of taxonomic literature enabling the identification of parasitoid genera is now available and relatively accessible to ecologists without specialised taxonomic training. The aim of this section of the current study was therefore to use these taxonomic resources and access to specialised taxonomic expertise, particularly in the Natural History Museum, London, to increase the level of taxonomic resolution for the parasitoid Hymenoptera being collected from Irish agricultural grasslands. Taxa were identified to genus level, and occasionally to species level whenever access to specialised taxonomist expertise permitted.

2.2 Methods

The parasitoid Hymenoptera collected from ongoing Ag-Biota monitoring and studies reported in Chapters 3 and 4 of the current report were systematically identified to genus level using the taxonomic literature listed in Table 2.1. Visits to the Natural History Museum, London, by the first author provided the necessary training to use these keys. The following specialists kindly provided assistance with confirmation of all generic identifications made: Dr Gavin Broad, Dr Andrew Polaszek, Dr David Notton and Dr John Noyes of the Natural History Museum, London, and Mr Hannes Baur of the Natural History Museum, Bern, Switzerland. In some instances, and with the help of the above specialists, it was possible to identify a small number of taxa to species level. In turn, the first author of this report provided subsequent taxonomic training to Mr Stephen McCormack, a PhD student funded by an ERTDI Doctoral Scholarship (2005-PHD-21) attached to the Ag-Biota research group.

2.3 Results and Discussion

A total of 27,206 individuals representing 178 genera of parasitoid Hymenoptera were collected and identified during the course of the project. A complete list of the taxa identified is given in Table 2.2, including species identifications where this was possible. The list includes ten taxa not previously recorded from Ireland. All of these new records for the Irish fauna were collected from moderately-to-intensively managed agricultural grasslands, suggesting that there may be much Irish biodiversity yet to be discovered, even in the most widespread and potentially undervalued habitat in the Irish landscape.

Table 2.1. Taxonomic keys used in the identification of parasitoid Hymenoptera from Irish grasslands.

Family	Subfamily	Author
Aphelinidae	–	Woolley, 1997
Braconidae	Alysiinae	Wharton, 1997a
Braconidae	Aphidiinae	Achterberg, 1997
Braconidae	Blaciinae	Sharkey, 1997
Braconidae	Cheloninae	Shaw, 1997a
Braconidae	Euphorinae	Shaw, 1997b
Braconidae	Macrocentrinae	Wharton, 1997b
Braconidae	Meteorinae	Shaw, 1997c
Braconidae	Microgastrinae	Whitfield, 1997
Braconidae	Opiinae	Wharton, 1997c
Braconidae	Rogadinae	Shaw, 1997d
Ceraphronidae	–	Alekseev, 1987
Cynipidae	Cynipiinae	Eady and Quinlan, 1963
Diapriidae	Belytinae	Nixon, 1957
Diapriidae	Diapriinae	Nixon, 1980
Dryinidae	–	Tryapitsyn, 1987
Encyrtidae	–	Noyes <i>et al.</i> , 1997
Eulophidae	Entodontinae	Schauff <i>et al.</i> , 1997
Eulophidae	Euderinae	Askew, 1968a
Eulophidae	Eulophinae	Askew, 1968a
Eulophidae	Tetrastichinae	Graham, 1987; Schauff <i>et al.</i> , 1997
Eurytomidae	–	Di Giulio, 1997; Nikol'skaya, 1952
Figitidae	Anacharitinae	Fergusson, 1986
Figitidae	Charapinae	Fergusson, 1986
Figitidae	Eucoilinae	Quinlan, 1978
Figitidae	Figitinae	Fergusson, 1986
Ichneumonidae	Adelognathinae	Townes, 1969
Ichneumonidae	Banchinae	Townes, 1970b
Ichneumonidae	Campopleginae	Townes, 1970b
Ichneumonidae	Cryptinae	Townes, 1970a
Ichneumonidae	Ctenopelmatinae	Townes, 1970b
Ichneumonidae	Diplazontinae	Fitton and Rotheray, 1982
Ichneumonidae	Ichneumoninae	Perkins and Eady, 1960
Ichneumonidae	Mesochorinae	Townes, 1971
Ichneumonidae	Microleptinae	Townes, 1971
Ichneumonidae	Orthocentrinae	Townes, 1971
Ichneumonidae	Pimplinae	Fitton <i>et al.</i> , 1988
Ichneumonidae	Tersiloschinae	Townes, 1971
Ichneumonidae	Tryphoninae	Townes, 1969
Megaspilidae	–	Alekseev, 1987; Fergusson, 1980
Mymaridae	–	Huber, 1997
Platygastridae	–	Kozlov, 1987
Proctotrupidae	–	Kozlov, 1987
Pteromalidae	–	Bouček and Rasplus, 1991
Scelionidae	–	Masner, 1980
Tetracampidae	–	Bouček and Heydon, 1997
Trichogrammatidae	–	Pinto, 1997

Table 2.2. A complete list of all parasitoid Hymenoptera taxa recorded in samples from Irish agricultural grasslands (new Irish records are marked in bold type).

Family	Subfamily	Genus and species
Aphelinidae	Apheliniinae	<i>Aphelinus</i> Dalman
Aphelinidae	Apheliniinae	<i>Centrodora</i> Förster
Braconidae	Alysiinae	<i>Chorebus</i> Haliday
Braconidae	Alysiinae	<i>Alloea</i> Haliday
Braconidae	Alysiinae	<i>Alysia</i> Latreille
Braconidae	Alysiinae	<i>Aphaereta</i> Foerster
Braconidae	Alysiinae	<i>Asobara</i> Foerster
Braconidae	Alysiinae	<i>Aspilota</i> Foerster
Braconidae	Alysiinae	<i>Chaenusa</i> Haliday
Braconidae	Alysiinae	<i>Chasmodon</i> Haliday
Braconidae	Alysiinae	<i>Coelinidea</i> Viereck
Braconidae	Alysiinae	<i>Dacnusa</i> Haliday
Braconidae	Alysiinae	<i>Dapsilarthra</i> Foerster
Braconidae	Alysiinae	<i>Dinotrema speculum</i> (Haliday)
Braconidae	Alysiinae	<i>Dinotrema</i> Foerster (other)
Braconidae	Alysiinae	<i>Orthostigma</i> Ratzeburg
Braconidae	Alysiinae	<i>Pentapleura</i> Foerster
Braconidae	Alysiinae	<i>Phaenocarpa</i> Foerster
Braconidae	Alysiinae	<i>Tanycarpa</i> Foerster
Braconidae	Aphidiinae	<i>Aphidius</i> Nees
Braconidae	Aphidiinae	<i>Binodoxys</i> Mackauer
Braconidae	Aphidiinae	<i>Diaeretiellus ephippium</i> (Haliday)
Braconidae	Aphidiinae	<i>Diaeretiella rapae</i> (McIntosh)
Braconidae	Aphidiinae	<i>Diaeretellus</i> Starý (other)
Braconidae	Aphidiinae	<i>Ephedrus</i> Haliday
Braconidae	Aphidiinae	<i>Monoctonus</i> Haliday
Braconidae	Aphidiinae	<i>Praon</i> Haliday
Braconidae	Aphidiinae	<i>Trioxy</i> Haliday
Braconidae	Blaciinae	<i>Blacus ambulans</i> Haliday
Braconidae	Blaciinae	<i>Blacus</i> Nees (other)
Braconidae	Cheloninae	<i>Chelonus</i> Panzer
Braconidae	Euphorinae	<i>Centistes</i> Haliday
Braconidae	Euphorinae	<i>Perilitus</i> Nees
Braconidae	Euphorinae	<i>Peristenus</i> Foerster
Braconidae	Macrocentrinae	<i>Macrocentrus</i> Curtis
Braconidae	Meteorinae	<i>Meteorus</i> Haliday
Braconidae	Microgastrinae	<i>Apanteles</i> Foerster
Braconidae	Microgastrinae	<i>Cotesia</i> Cameron
Braconidae	Microgastrinae	<i>Glyptapanteles</i> Ashmead
Braconidae	Microgastrinae	<i>Microplitis</i> Foerster
Braconidae	Microgastrinae	<i>Sathon fulcatus</i> Mason

Table 2.2 *contd.*

Family	Subfamily	Genus and species
Braconidae	Opiinae	<i>Opius</i> Wesmael
Braconidae	Rogadinae	<i>Aleiodes</i> Wesmael
Ceraphronidae	–	<i>Aphanogmus</i> Thomson
Ceraphronidae	–	<i>Ceraphron</i> Jurine
Cynipidae	Cynipiinae	<i>Aulacidea</i> Ashmead
Diapriidae	Belytinae	<i>Aclista</i> Foerster
Diapriidae	Belytinae	<i>Belyta</i> Jurine
Diapriidae	Belytinae	<i>Cinetus</i> Jurine
Diapriidae	Belytinae	<i>Opazon</i> Haliday
Diapriidae	Belytinae	<i>Pamis</i> Nixon
Diapriidae	Belytinae	<i>Paroxylabis</i> Kieffer
Diapriidae	Diapriinae	<i>Basalys</i> Westwood
Diapriidae	Diapriinae	<i>Basalys orion</i> Nixon
Diapriidae	Diapriinae	<i>Diapria</i> Latreille
Diapriidae	Diapriinae	<i>Idiotypa</i> Foerster
Diapriidae	Diapriinae	<i>Paramesius</i> Westwood
Diapriidae	Diapriinae	<i>Trichopria</i> Ashmead
Dryinidae	–	<i>Anteon</i> Jurine
Dryiniidae	–	<i>Gonatopus</i> Ljungh
Encyrtidae	Encyrtinae	<i>Copidosma</i> Ratzeburg
Encyrtidae	Encyrtinae	<i>Lamennaisia</i> Mercet
Encyrtidae	Encyrtinae	<i>Syrphophagous</i> Ashmead
Encyrtidae	Tetracneminae	<i>Anagyrus</i> Howard
Encyrtidae	Tetracneminae	<i>Rhopus</i> Förster
Encyrtidae	Tetracneminae	<i>Rhopus acaetes</i> Walker
Eulophidae	Entodontinae	<i>Asecodes</i> Nees
Eulophidae	Entodontinae	<i>Chrysocharis</i> Förster
Eulophidae	Entodontinae	<i>Closterocerus</i> Westwood
Eulophidae	Entodontinae	<i>Holcopelte</i> Förster
Eulophidae	Entodontinae	<i>Ionympha</i> Walker
Eulophidae	Entodontinae	<i>Omphale</i> Haliday
Eulophidae	Entodontinae	<i>Pediobius</i> Walker
Eulophidae	Euderinae	<i>Euderus</i> Haliday
Eulophidae	Eulophinae	<i>Diglyphus</i> Walker
Eulophidae	Eulophinae	<i>Eulophus</i> Walker
Eulophidae	Eulophinae	<i>Hemiptarsenus ornatus</i> (Nees)
Eulophidae	Eulophinae	<i>Hemiptarsenus unguicellus</i> (Zetterstedt)
Eulophidae	Eulophinae	<i>Hemiptarsenus fulvicollis</i> Westwood
Eulophidae	Eulophinae	<i>Necremnus</i> Thomson
Eulophidae	Eulophinae	<i>Pnigalio</i> Schrank
Eulophidae	Tetrastichinae	<i>Aprostocetus</i> sp. 1 Westwood

Table 2.2 contd.

Family	Subfamily	Genus and species
Eulophidae	Tetrastichinae	<i>Aprostocetus</i> Westwood (<i>other</i>)
Eurytomidae	Eurytominae	<i>Eurytoma</i> Illiger
Eurytomidae	Eurytominae	<i>Tetramesa</i> Walker
Figitidae	Anacharitinae	<i>Anacharis</i> Dalman
Figitidae	Charapinae	<i>Alloxysta abdera</i> Fergusson
Figitidae	Charapinae	<i>Alloxysta brachyptera</i> (Hartig)
Figitidae	Charapinae	<i>Alloxysta brevis</i> (Thomson)
Figitidae	Charapinae	<i>Alloxysta macrophadna</i> (Hartig)
Figitidae	Charapinae	<i>Phaenoglyphis villosa</i> (Hartig)
Figitidae	Eucoilinae	<i>Kleidotoma</i> Westwood
Figitidae	Eucoilinae	<i>Tribliographa</i> Förster
Figitidae	Eucoilinae	<i>Rhoptromeris</i> Förster
Figitidae	Figitinae	<i>Melanips</i> Haliday
Figitidae	Figitinae	<i>Sarothrus tibialis</i> (Zetterstedt)
Ichneumonidae	Adelognathinae	<i>Adelognathus dorsalis</i> (Gravenhorst)
Ichneumonidae	Banchinae	<i>Lissonota</i> Gravenhorst
Ichneumonidae	Campopleginae	<i>Campoletis</i> Förster
Ichneumonidae	Campopleginae	<i>Diadegma</i> Förster
Ichneumonidae	Cryptinae	<i>Aclastus minutus</i> (Bridgman)
Ichneumonidae	Cryptinae	<i>Aclastus solutus</i> (Thomson)
Ichneumonidae	Cryptinae	<i>Aclastus</i> Förster (<i>other</i>)
Ichneumonidae	Cryptinae	<i>Aritranis director</i> (Thunberg)
Ichneumonidae	Cryptinae	<i>Atractodes</i> Gravenhorst
Ichneumonidae	Cryptinae	<i>Endasys</i> Förster
Ichneumonidae	Cryptinae	<i>Gambrus</i> Förster
Ichneumonidae	Cryptinae	<i>Gelis</i> Thunberg
Ichneumonidae	Cryptinae	<i>Oecotelma</i> Townes ¹
Ichneumonidae	Cryptinae	<i>Phygadeuon</i> Gravenhorst
Ichneumonidae	Cryptinae	<i>Stibeutes</i> Förster
Ichneumonidae	Cryptinae	<i>Stilpnus</i> Gravenhorst
Ichneumonidae	Ctenopelmatinae	<i>Campodorus</i> Förster
Ichneumonidae	Ctenopelmatinae	<i>Perilissus</i> Förster
Ichneumonidae	Diplazontinae	<i>Diplazon</i> Nees
Ichneumonidae	Diplazontinae	<i>Promethes sulcator</i> (Gravenhorst)
Ichneumonidae	Diplazontinae	<i>Sussaba dorsalis</i> (Holmgren)
Ichneumonidae	Diplazontinae	<i>Syrphoctonus</i> Förster
Ichneumonidae	Ichneumoninae	<i>Barichneumon</i> Thomson
Ichneumonidae	Ichneumoninae	<i>Epitomus infuscatus</i> (Gravenhorst)
Ichneumonidae	Ichneumoninae	<i>Limerodops elongates</i> (Brischke)
Ichneumonidae	Ichneumoninae	<i>Amblyteles</i> Wesmael
Ichneumonidae	Mesochorinae	<i>Mesochorus</i> Gravenhorst

Table 2.2 contd.

Family	Subfamily	Genus and species
Ichneumonidae	Microleptinae	<i>Microleptes</i> Gravenhorst
Ichneumonidae	Orthocentrinae	<i>Orthocentrus</i> Gravenhorst
Ichneumonidae	Orthocentrinae	<i>Plectiscidea</i> Viereck
Ichneumonidae	Orthocentrinae	<i>Stenomacrus</i> Förster
Ichneumonidae	Pimplinae	<i>Tromatobia variabilis</i> (Holmgren)
Ichneumonidae	Tersiloschinae	<i>Barycnemis</i> Förster
Ichneumonidae	Tersiloschinae	<i>Microdiaparsis neoversutus</i> Horstmann
Ichneumonidae	Tryphoninae	<i>Eridolius</i> Förster
Ichneumonidae	Tryphoninae	<i>Polyblastus</i> Hartig
Megaspilidae	Megaspilinae	<i>Conostigmus</i> Dahlbom
Megaspilidae	Lagynodinae	<i>Lagynodes</i> Foerster
Megaspilidae	Megaspilinae	<i>Dendrocerus aphidum</i> (Rondani)
Megaspilidae	Megaspilinae	<i>Dendrocerus carpenteri</i> (Curtis)
Megaspilidae	Megaspilinae	<i>Dendrocerus dubiosus</i> (Ratzeburg)
Megaspilidae	Megaspilinae	<i>Dendrocerus laticeps</i> (Hedricke)
Mymaridae	–	<i>Alaptus</i> Westwood
Mymaridae	–	<i>Anagrus</i> Haliday
Mymaridae	–	<i>Anaphes</i> Haliday
Mymaridae	–	<i>Clereuchus</i> Enock
Mymaridae	–	<i>Erythmelus</i> Enock
Mymaridae	–	<i>Gonatocerus</i> Nees
Mymaridae	–	<i>Litus</i> Haliday
Mymaridae	–	<i>Mymar</i> Curtis
Mymaridae	–	<i>Ooctonus</i> Haliday
Mymaridae	–	<i>Polynema</i> Haliday
Platygastridae	Platygastrinae	<i>Inostemma</i> Haliday
Platygastridae	Platygastrinae	<i>Leptacis</i> Foerster
Platygastridae	Platygastrinae	<i>Platygaster</i> Latreille
Platygastridae	Platygastrinae	<i>Platygaster B</i> Latreille
Platygastridae	Platygastrinae	<i>Synopeas</i> Foerster
Proctotrupidae	–	<i>Codrus</i> Panzer
Proctotrupidae	–	<i>Phaenoserphus</i> Pschorn-Walcher
Proctotrupidae	–	<i>Phaenoserphus</i> Kieffer
Pteromalidae	Asaphinae	<i>Asaphes suspensis</i> (Nees)
Pteromalidae	Asaphinae	<i>Asaphes vulgaris</i> Walker
Pteromalidae	Microgastrinae	<i>Coruna</i> Walker
Pteromalidae	Microgastrinae	<i>Halticoptera aenea</i> (Walker)
Pteromalidae	Miscogasterinae	<i>Halticoptera circulus</i> (Walker)
Pteromalidae	Miscogasterinae	<i>Merismus rufipes</i> Walker
Pteromalidae	Miscogasterinae	<i>Miscogaster rmaculata</i> Walker
Pteromalidae	Miscogasterinae	<i>Rhcnocoelia</i> Graham

Table 2.2 *contd.*

Family	Subfamily	Genus and species
Pteromalidae	Miscogasterinae	<i>Seladerma</i> Walker
Pteromalidae	Miscogasterinae	<i>Thinodytes cyzicus</i> (Walker)
Pteromalidae	Ormocerinae	<i>Semiotellus</i> Westwood
Pteromalidae	Panstenoninae	<i>Panstenon oxylus</i> (Walker)
Pteromalidae	Pireninae	<i>Gastroncistrus</i> Westwood
Pteromalidae	Pireninae	<i>Macroglenes</i> Westwood
Pteromalidae	Pteromalinae	<i>Callitula pyrrhogaster</i> (Walker)
Pteromalidae	Pteromalinae	<i>Callitula</i> Spinola (<i>other</i>)
Pteromalidae	Pteromalinae	<i>Chlorocytus formosus</i> (Walker)
Pteromalidae	Pteromalinae	<i>Cyrtogaster vulgaris</i> Walker
Pteromalidae	Pteromalinae	<i>Cyrtogaster</i> Walker (<i>other</i>)
Pteromalidae	Pteromalinae	<i>Homoporus febriculosus</i> (Girault)
Pteromalidae	Pteromalinae	<i>Homoporus luniger</i> (Nees)
Pteromalidae	Pteromalinae	<i>Meraporus graminicola</i> (Walker)
Pteromalidae	Pteromalinae	<i>Mesopolobus aequus</i> (Walker)
Pteromalidae	Pteromalinae	<i>Mesopolobus diffinis</i> (Walker)
Pteromalidae	Pteromalinae	<i>Mesopolobus laticornis</i> (Walker)
Pteromalidae	Pteromalinae	<i>Mesopolobus</i> Westwood (<i>other</i>)
Pteromalidae	Pteromalinae	<i>Pachyneuron</i> Walker
Pteromalidae	Pteromalinae	<i>Pteromalus</i> Swederus
Pteromalidae	Pteromalinae	<i>Spaniopus dissimilis</i> Walker
Pteromalidae	Pteromalinae	<i>Spaniopus</i> Walker (<i>other</i>)
Pteromalidae	Pteromalinae	<i>Spintherus dubius</i> (Nees)
Pteromalidae	Pteromalinae	<i>Stenomalina communis</i> cf (Nees)
Pteromalidae	Pteromalinae	<i>Toxeuma fuscicorne</i> Walker
Pteromalidae	Pteromalinae	<i>Trichomalopsis hemiptera</i> (Walker)
Pteromalidae	Pteromalinae	<i>Trichomalopsis</i> Crawford (<i>other</i>)
Pteromalidae	Pteromalinae	<i>Trichomalus campestris</i> (Walker)
Pteromalidae	Pteromalinae	<i>Trichomalus rufinus</i> (Walker)
Pteromalidae	Pteromalinae	<i>Trichomalus</i> Thomson (<i>other</i>)
Pteromalidae	Spalangiinae	<i>Spalangia</i> Latreille
Scelionidae	Scelioninae	<i>Baeus</i> Haliday
Scelionidae	Teleasinae	<i>Trimorus</i> Foerster
Scelionidae	Telenominae	<i>Telenomus</i> Haliday
Tetracampidae	—	<i>Epiclerus</i> Haliday
Trichogrammatidae	—	<i>Oligosita</i> Walker
Trichogrammatidae	—	<i>Trichogramma</i> Westwood

¹Awaiting confirmation of identification.

3 The Potential of Parasitoid Hymenoptera as Bioindicators of the Diversity of Other Arthropods

3.1 Background

Following the signing of the CBD, which agreed to integrate biodiversity policy into all economic sectors (UNEP, 1992), the European Commission (2000, 2001) published a Biodiversity Action Plan (BAP) for Agriculture (COM(2001)162 vol. III) as part of a strategy to halt the global decline in biodiversity by 2010. Amongst the recommendations of a subsequent meeting of the European Platform for Biodiversity Research Strategy (EPBRS) in Ireland in 2004, there was agreement on the urgent need to develop monitoring systems to evaluate the performance of the Common Agricultural Policy (CAP) in terms of halting biodiversity loss (EPBRS, 2004). Measuring biodiversity in its entirety is impossible because of the vast numbers of species of living organisms and the manpower that would be required for their collective identification. There is therefore a need for the identification of appropriate bioindicators, taxa that can be selected and monitored on the basis of their being representative of wider biodiversity (Duelli and Obrist, 2003a; New, 2005). McGeoch (1998) defined a biological indicator as: *"a species, or group of species that readily reflects: the abiotic or biotic state of an environment; represents the impact of environmental change on a habitat, community or ecosystem; or is indicative of the diversity of a subset of taxa, or of wholesale diversity within an area"*. There can be no single indicator for biodiversity in all contexts. The selection of appropriate bioindicators is likely to depend on the dimension of the wider environment being evaluated (e.g. the habitat type, or economic sector) and is usually guided by issues such as the availability of taxonomic expertise and resources (Duelli and Obrist, 2003b). An essential first step in selecting useful biodiversity indicators is to find groups whose incidence correlates well with overall taxon richness in a particular context (Sauberer *et al.*, 2004).

In order for bioindicators to be used to their fullest advantage, it is also necessary to understand the ecological relationships between the chosen indicator group(s) and wider community structure, and the particular ecological influences they reflect (Paoletti, 1999).

Following an initial programme of monitoring by the Ag-Biota project at ten farm sites, evidence was found to support the original hypothesis that parasitoid wasps (identified only to family level) were the insect group with greatest potential as bioindicators of wider arthropod diversity (Fig. 3.1). We wished to test the hypothesis that, because of their close ecological relationships with practically all other insect groups (Quicke, 1997), the Hymenoptera Parasitica have good theoretical potential as functionally significant bioindicators within agro-ecosystems (LaSalle, 1993). This part of the project began with a simple hypothesis that parasitoid wasps would be good indicators of arthropod diversity in agricultural grasslands. When this was supported by data collected from an initial ten monitoring sites, the hypothesis was further tested at a higher level of taxonomic resolution on a much larger scale of 50 commercial farm sites.

3.2 Aims

- To validate the relationship between parasitoid wasp diversity and the diversity of other arthropod taxa collected in sward samples from the original ten Ag-Biota farm sites when the level of parasitoid taxonomic resolution is increased from family to genus level.
- To further test the relationship between parasitoid wasp diversity and the diversity of other arthropods using an independent, and much larger, data set collected from agricultural grasslands at an additional 50 farm sites.

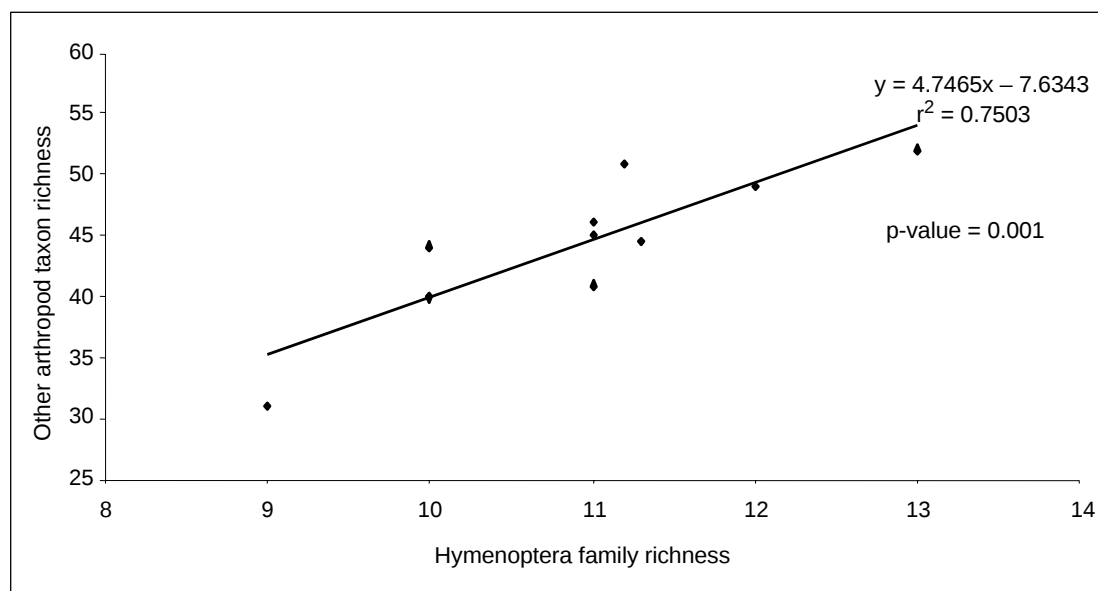


Figure 3.1. The relationship between hymenopteran parasitoid family richness and the total taxon richness of all other arthropods (excluding parasitoid Hymenoptera) collected in Vortis suction samples from agricultural grassland swards at ten initial Ag-Biota monitoring sites.

3.3 Methods

3.3.1 Testing parasitoids as bioindicators for wider arthropod diversity using data from the initial Ag-Biota monitoring sites

3.3.1.1 Site selection

Ten individual grassland fields situated on farms representative of a wide range of farm management intensities were sampled across south-east Ireland in 2002. The fields for this initial monitoring work were selected at five paired locations: four field pairs were situated on Teagasc research centres at Solohead, Grange, Oak Park and Johnstown Castle and on nearby commercial farms under similar farming management regimes. The fifth pair of fields, each of different management intensity, was situated on the UCD research farm at Lyons Estate in Co. Kildare (Table 3.1).

3.3.1.2 Arthropod sampling

Vegetation arthropods were sampled from randomly chosen grassland fields at each farm site using a Vortis Insect Suction Sampler (Burkard Manufacturing Co. Ltd, Rickmansworth, Hertfordshire, UK) (Arnold, 1994). Ten samples were collected from each field. Each sample consisted of the pooled catch from three

randomly selected sampling spots, individually sampled for 10 s. The total area sampled in each field was 0.6 m². All samples were collected during August 2002 in dry-weather conditions between 11:00 h and 15:00 h. Where sites were being rotationally grazed, samples were taken at the mid-point in sward recovery (medium sward length) within the grazing cycle. The catches were preserved in 70% ethanol prior to sorting and identification. Five major arthropod groups dominated the collected samples: Hemiptera, Araneae and Coleoptera were identified to species level, Diptera were identified to family level and parasitic Hymenoptera were initially identified to family level and, subsequently, as part of the current study to genus level.

3.3.1.3 Data analysis

After pooling all samples from individual fields, and so using sites as replicates, the relationship between the taxon richness of each major arthropod group, in turn, and the total taxon richness of all other arthropod groups (excluding the group being evaluated – see Sauberer *et al.* (2004) was determined by linear model analysis using the R statistical package, version 2.4.1 (R Core Development Team, 2006).

Table 3.1. Location, farming type and management system at the initial ten sampled farm sites. Grid references are not given for the commercial farms to protect farmer confidentiality.

County	Location	Grid Reference	Farming type	Management
Wexford	Teagasc, Johnstown Castle (JC)	T026166	Mixed grassland	Intensive rotational dairy grassland
Wexford	Johnstown Castle, commercial (JCC)		Dairy	Intensive rotational dairy grassland
Tipperary	Teagasc, Solohead (SH)	R863395	Dairy	Intensive rotational dairy grassland
Tipperary	Solohead, commercial (SHC)		Dairy	Intensive rotational dairy grassland
Carlow	Teagasc, Oak Park (OP)	S731902	Mainly arable	Extensive semi-continuous grazing
Carlow	Oak Park, commercial (OPC)		Mainly arable	Extensive semi-continuous parkland
Meath	Teagasc, Grange (G)	N884530	Beef grassland	REPS rotational suckler beef
Meath	Grange, commercial (GC)		Dry stock grassland	REPS semi-continuous beef/sheep grazing
Kildare	UCD, Lyons Dairy (LD)	N976283	Mixed livestock/arable	Intensive
Kildare	UCD, Lyons Hill (LH)	N976287	Mixed livestock/arable	Extensive

3.3.2 *Testing parasitoids as bioindicators for wider arthropod diversity using data from an extended Ag-Biota survey of 50 commercial farm sites*

3.3.2.1 *Site selection*

Fifty commercial farms were randomly chosen for an extended survey study in south-east Ireland using the National Farm Survey (NFS) database to select a random sample of farms. The NFS maintains a database of Irish farm statistics by regular collation of data from a nationally representative sample of over 1,000 Irish farms (Connolly *et al.*, 2004). A random subsample of 50 predominantly grassland farms in south-east Ireland, proportionately stratified by county (Carlow, Cork, Kilkenny, Laois, Meath, Waterford, Wexford and Wicklow) and main livestock farm enterprise (dairying, beef cattle, and sheep), was selected from this database with the assistance of NFS staff.

3.3.2.2 *Arthropod sampling*

Vegetation arthropods were sampled from a randomly chosen, representative grassland pasture at each farm site in July 2005, using a Vortis Insect Suction Sampler (Burkard Manufacturing Co. Ltd, Rickmansworth, Hertfordshire, UK) (Arnold, 1994). Based on the experience gained from sampling at the original ten Ag-Biota sites, the number of Vortis samples collected

from each field was increased from ten to 20, with each sample consisting of the pooled catch from six (increased from three) randomly selected sampling spots individually sampled for 10 s. The total area sampled within each field was therefore increased from 0.6 m² to 2.4 m². This was effectively a fourfold increase in sampling intensity compared with the original ten-site study. Other sampling details were similar to those given in Section 3.3.1.2.

3.3.2.3 *Data analysis*

After pooling all samples from individual sites, and so using sites as replicates, the relationship between the taxon richness of each major arthropod group, in turn, and the total taxon richness of all other arthropod groups was investigated using linear model analysis as described in Section 3.3.1.3. Additionally, the substantially larger samples collected in the 50-site survey enabled analysis of the relationship between total abundance of parasitoid wasps (total numbers of individuals) and the diversity (taxon richness) of all other arthropod groups.

3.4 Results

3.4.1 *Initial ten-site study*

Seventy-five parasitoid wasp genera, representing 16 families, were identified in the total sample of 2,054 individuals collected in the initial ten-site survey. This

total parasitoid fauna was collected from an effective total pooled area of 6 m² of sampled grassland. A wide range of other arthropod taxa was also collected and identified in this ten-site survey (Table 3.2).

When relationships between the taxon richness of individual major arthropod groups and total overall arthropod taxon richness (excluding the evaluated group) were investigated, only the parasitoid Hymenoptera, identified to either family or genus level, showed a potentially useful significant relationship, although the relationship at genus level was weaker than that for family (Table 3.3). When making multiple simultaneous assessments of the significance of individual correlation statistics, a Bonferroni adjustment of the critical probability value (dividing the conventionally accepted 5% error by the number of relationships being simultaneously tested) is normally advocated to guard against the multiplied risk of error (Sauberer *et al.*, 2004). However this adjustment is extremely conservative, and when routinely applied in the assessment of significance, can increase the risk of a Type II error, whereby a genuinely significant finding is rejected as not significant. We therefore chose to interpret the data from the initial ten-site study

as providing some support for our original hypothesis that the diversity of parasitoid Hymenoptera is a good indicator of wider arthropod diversity, and to seek independent verification of this relationship from the data collected in the extended monitoring of grassland arthropod populations in the 50-site survey.

3.4.2 Extended 50-site study

As the conclusion from the initial ten-site study regarding our hypothesis concerning the bioindicator value of parasitoid wasps was encouraging but not entirely conclusive, independent verification of the hypothesised relationship was sought from data provided by the more extensive 50-farm survey. With an increased number of sampled sites, and a substantially increased sample size from each surveyed field, analyses showed that the taxon diversity of all major arthropod groups was individually, significantly correlated with the total taxon richness of all other groups (Table 3.4).

With the exception of the Order Diptera, the statistical significance of these relationships in the larger 50-site data set is maintained even after Bonferroni adjustment of the acceptable probability of error. However, the r^2 values between the incidence of

Table 3.2. Summary of the total numbers of arthropod taxa collected from a total sampled area of 6 m² of agricultural grassland in the initial ten-site survey.

Group	No. families	No. genera	No. species	No. individuals
Coleoptera	9	42	70	720
Diptera	25	–	–	3975
Hemiptera	5	12	16	2110
Hymenoptera	16	75	–	2,054
Araneae	4	10	13	2,119

Table 3.3. Summary of regression statistics for the relationships between taxon richness of individual arthropod groups and the total taxon richness of all other arthropod groups using data from the initial ten-site study.

Major group (level of taxonomic resolution)	r^2	p-value*
Coleoptera (species)	0.1175	0.225
Diptera (families)	0.2946	0.409
Hemiptera (species)	0.0086	0.798
Hymenoptera (genera)	0.4542	0.033
Hymenoptera (families)	0.7503	0.001
Hymenoptera (abundance of individuals)	0.062	0.488
Araneae (species)	0.0490	0.536

*Bonferroni-adjusted critical p-value = 0.05/6 = **0.0083**

Table 3.4. Summary of regression statistics for the relationship between taxon richness of individual arthropod groups (and the abundance of parasitoid wasps), and the total taxon richness of all other arthropod groups using data from the 50-site survey.

Major group (level of taxonomic resolution)	r^2	p-value*
Coleoptera (species)	0.1545	0.006
Diptera (family)	0.0960	0.032
Hemiptera (species)	0.2849	>0.0001
Hymenoptera (family)	0.1929	0.001
Hymenoptera (genus)	0.4247	>0.0001
Hymenoptera (abundance of individuals)	0.5006	>0.0001
Araneae (species)	0.1573	0.005

*Bonferroni-adjusted critical p-value = $0.05/7 = 0.0074$.

parasitoid Hymenoptera (in terms of numbers of genera, or individual wasps) and total arthropod taxon richness were stronger than for any other individual group, r^2 values of 0.4247 and 0.5006 for genera and individuals, respectively, compared with a 'next best' r^2 of 0.2849 for the taxon richness of Hemiptera (Table 3.4).

3.5 Discussion

Analysis of the relationship between parasitoid wasp diversity and the diversity of all other arthropod taxa using data provided by the initial study of just ten paired grassland sites, provided some evidence to support our original hypothesis regarding the potential bioindicator value of this specialised group of insects. Somewhat unexpectedly, however, increasing the level of taxonomic resolution from families to genera in the analysis of this original data set did not improve the observed relationship ($r^2 = 0.7503$ and 0.4542 for parasitoid families and genera, respectively). Both the numbers of sites and the size of samples taken in this initial study were rather small, making it necessary to treat this result with caution. As a relatively very large total number of (75) parasitoid genera were collected in the relatively small pooled sample, totalling only 6 m^2 of grassland ($0.6 \text{ m}^2 \times 10$ sites), it is likely that the actual collection of any individual genus or parasitoid abundance in this initial study was subject to a high degree of uncertainty and randomness. In these circumstances, it might be expected that the total numbers of parasitoid families collected at each site would be more complete and consistent, and should therefore show a stronger correlation with total observed arthropod diversity.

The sample size collected from individual fields was increased to 2.4 m^2 in the larger study of 50 grassland sites. Analysis of this data set showed that the individual taxon richness of all major arthropod groups, except Diptera, was correlated with total overall arthropod diversity. This relationship was strongest, however, for the parasitoid Hymenoptera, identified to genus, but the total abundance of parasitoid wasps was found to be even more strongly related to total observed arthropod diversity. Whilst a degree of caution is again advisable when interpreting a correlation from a single data set, this observation may be particularly significant. Despite the improving level of taxonomic knowledge and literature to facilitate the identification of parasitoid wasps, such work will always require a relatively high level of expertise, and so be logistically difficult to integrate into any realistic framework of widespread monitoring. The sampling and quantification of total parasitoid abundance (total numbers of individuals), however, would be a relatively straightforward and practicable option for routine monitoring. Further analyses to test the relationship between parasitoid abundance (both numbers of individuals and taxon richness) and total arthropod diversity using independent data sets collected from the grassland field experiments by the Ag-Biota project is ongoing. Should this further work confirm the potentially extremely useful relationship between simple numbers of parasitoid Hymenoptera and overall arthropod diversity, there is a very reasonable prospect that parasitoid wasps can be used as a bioindicator group for routine assessment of wider arthropod diversity, at least within the context of agricultural grasslands, which comprise the greater

proportion of the Irish countryside. It is likely that this relationship may hold in other agro-ecosystems (including cereals and potential new biofuel crops), making the parasitic Hymenoptera a more widely useful bioindicator group for agro-ecosystems. However, this relationship needs to be tested in other

crop habitats. In the meantime, the current project has gone on to investigate and quantify the community structure of parasitoid populations within different agricultural grassland contexts and to use this knowledge to develop a better understanding of the ecological effects of farm practice.

4 Diversity and Ecology of Parasitoid Wasps in a Manipulated Grassland Field Margin Experiment

4.1 Background

The intensification of grassland management, brought about by increases in grazing intensity, fertiliser usage and other chemical inputs, is widely regarded as having led to widespread landscape degradation and a loss of biodiversity (McLaughlin and Mineau, 1995; Duelli, 1997; Buchs, 2003; Hoffman and Greef, 2003). From an agri-environmental perspective, it is therefore important to understand how changing grassland husbandry practices impact on biological diversity. Maintenance of biodiversity in agro-ecosystems is not only important from a conservation perspective, but has significant and direct functional benefits to the farming industry ensuring the greater effectiveness of pollination, organic matter decomposition and natural control of pest populations, leading to greater ecological stability and sustainability (LaSalle and Gauld, 1993; Paoletti, 1995, 1999; New, 2005). There is therefore a need to develop agricultural systems that protect and enhance biodiversity in order to ensure longer-term sustainability (Paoletti, 1995; Buchs, 2003), as well as promoting the conservation of natural populations. There is now a substantial literature regarding the functional benefits of maintaining conservation strips and field margins to enhance the diversity and abundance of beneficial predatory arthropods in intensively managed arable landscapes. Arable farming, by its very nature, is heavily input-driven (including the use of broad-spectrum pesticides) and subject to regular disturbance by soil cultivations. In contrast, grassland farm systems are often perceived as having less impact on arthropod populations, but the limited research available would suggest that this is not necessarily the case.

The reduction in botanical and plant structural (especially sward height) diversity associated with intensive grazing and silage cutting systems can have profoundly negative impacts on arthropod populations (Purvis and Curry, 1981; Foster *et al.*, 1997; Clausen *et al.*, 2001). The spreading of organic manures

(Desender, 1982) and pesticide applications (Longley and Sotherton, 1997; Asteraki *et al.*, 2004) are also known to have deleterious effects on grassland arthropods. Conservation field margins may also provide a useful strategy for the protection of biodiversity in grassland farming by ensuring the retention of plant species and structural diversity, avoiding the disturbance of intensive grazing and cutting cycles and the negative impacts of high inputs of fertilisers and manures (Lagerlof and Wallin, 1993; Longley and Sotherton, 1997; Denys and Tschardtke, 2002; Meek *et al.*, 2002; Asteraki *et al.*, 2004; Sheridan, 2005). Field margins in both arable and grassland farming can potentially contribute to the sustainability of agricultural systems by providing habitats for functionally important beneficial species and reducing the need for pesticide use (Marshall and Moonen, 2002). The Rural Environment Protection Scheme (REPS) includes measures to protect biodiversity within grassland field margins. These include the exclusion of agricultural inputs, including organic and inorganic fertilisers and pesticides (other than 'spot treatment' of proscribed weed species), from a 1.5-m wide strip adjacent to hedgerows, watercourses and other ecological features. Within the current variant of REPS 3 launched in 2006, Measure 2 requires the implementation of a sustainable grassland management plan that aims to protect habitat and minimise poaching, overgrazing and soil erosion. Where grassland is being reseeded, an untilled, unploughed and unsprayed margin of 1.5 m must be left in place. Measure 5 aims to conserve, maintain and enhance boundary fences, roadside fences, stone walls and hedgerows in the interest of stock control, animal health, wildlife and scenic appearance of the area (Department of Agriculture and Food, 2006). However, to date, very little research has been undertaken within Ireland to validate or improve the effectiveness of REPS measures relating to field margins, aside from one study that suggested that carabid beetle populations in

field margins did not benefit when participating and non-participating farms were compared (Feehan *et al.*, 2005).

Increased botanical and structural diversity of plants in field margins can potentially provide food and shelter for an increased number and diversity of parasitoid hosts. Studies have shown that increased plant species and structural diversity can enhance parasitoid diversity in agro-ecosystems (Altieri *et al.*, 1993). However, Menalled *et al.* (1999) found no relationship between landscape complexity and parasitoid diversity, or the rate of host parasitism by parasitoids. Important aspects of parasitoid biology are likely to be influenced by host biology, and so be reflected in their host range. It might, for example, be expected that parasitoid communities associated with well-concealed hosts (e.g. plant-miners) would be dominated by generalist parasitoids (e.g. idiobionts) as concealment provides protection for a defenceless parasitised host, whereas parasites of exposed hosts (e.g. aphids) would be more likely to be specialised koinobionts, which allow their hosts to continue their development, at least during the earlier stages of parasitoid development (Hawkins, 1994). The term 'parasitoid guild' has been used to define the many different possible ways in which parasitoids can interact with different stages in the life cycles of their hosts (Quicke, 1997). A host can be attacked by a number of different parasitoid guilds at different stages in its life cycle, and this number is influenced by many factors including the ecological, and particularly the trophic, niche (or niches) of the host (Quicke, 1997). Separating parasitoids into idiobionts (which kill or permanently paralyse their host when a female wasp oviposits in, or on it) and koinobionts (which allow continued development of the host during the early stages of immature parasitoid development) can be another potentially useful way to investigate and better understand host-parasitoid interactions (Hawkins, 1994).

The intricacies of parasitoid biologies, and the complex and specific associations of different parasitoid taxa with different host groups, mean that they have an indicator potential beyond their own abundance or total taxon diversity. Potentially, knowledge of parasitoid community structure, and in particular the relative

abundance of taxa with particular host ranges and developmental biologies, can provide an unrivalled ecological insight into the underlying influences of management practices on the wider diversity and community structure of arthropods within agro-ecosystems.

4.2 Aims

The aims of this part of the overall study were to quantify and analyse parasitoid community structures in a field margin experiment established in intensively managed agricultural grassland paddocks at Teagasc, Johnstown Castle, as a means to evaluate further the bioindicator potential of parasitoid Hymenoptera. Specific objectives were:

- To quantify the effects of the fenced margin treatments on the taxon richness and abundance of parasitoid Hymenoptera
- To further investigate the relationship between the abundance and taxon richness of parasitoid Hymenoptera and total arthropod diversity observed in the field margin treatments
- Using available published literature, to identify the host ranges of the parasitoids collected
- To quantify and analyse the community structure of parasitoid assemblages in the margin treatments
- Using knowledge of host ranges, to further explore and demonstrate the bioindicator potential of knowledge concerning parasitoid host ranges.

4.3 Methods

4.3.1 Experimental design

The field margin experiment was located on the dairy farm at the Teagasc Research Centre, Johnstown Castle, Co. Wexford (Grid Reference: T026166). All traditional field hedgerows and their associated herbaceous margins had been removed from this site during a process of grassland management intensification during the 1970s and 1980s. The result of this process was the subdivision of the site into intensively grazed paddocks dominated by *Lolium perenne* L. (perennial ryegrass), and fenced with electric fencing.

Prior to the start of, and during the experiment, the paddocks were grazed by a Friesian dairy herd on a standard 21-day rotation with each paddock cut once for silage in alternate years. In February 2002, a conservation field margin experiment was established by physically fencing off three replicate 90-m long strips of 1.5 m, 2.5 m and 3.5 m width at the edges of existing paddocks (total = 9 strips). Each strip was subsequently divided into three 30-m long plots to accommodate three different field margin treatments (a–c) in a fully randomised block design. The treatments were as follows:

- (a) *Fenced only* (FO): the existing grass sward within the strip simply fenced off from the adjacent paddock
- (b) *Rotavated and Fenced* (ROT): the existing vegetation removed with a glyphosate-based herbicide and the strip rotavated and left to revegetate naturally after being fenced off
- (c) *Rotavated, Reseeded and Fenced* (RS): the existing vegetation removed with glyphosate-based herbicide and the strip rotavated and reseeded with a grass and wildflower seed mixture before being fenced off.

Once established, these fenced margin treatments (a–c) were protected from all cattle grazing and normal nutrient inputs that continued as normal on the adjacent paddocks. *Unfenced control* (UC) margin strips (treatment d) of similar length (30 m) and width (1.5, 2.5 or 3.5 m) were established adjacent to each of the nine replicate blocks. These unfenced margins remained, in effect, part of the associated paddock, and continued to be grazed, fertilised and cut for silage in exactly the same intensive fashion as the remainder of the paddocks. Further details on the experimental design are given by Sheridan *et al.* (2003) and Sheridan (2005).

4.3.2 Arthropod sampling

Vegetation arthropods were sampled four times, in June and August 2004 and 2005, respectively, using a Vortis suction sampler (Burkard Manufacturing Co. Ltd, Rickmansworth, Hertfordshire, UK) (Arnold, 1994). An aggregate sample consisting of the pooled catch from ten randomly selected spots, each

individually sampled for 10 s duration, was collected from each margin strip, including the UC strips. The total area sampled per experimental strip was 0.19 m² on each sampling occasion. Catches were stored in 70% ethanol until identification. Parasitoids were identified using the keys listed in Table 2.1.

4.3.3 Data analysis

4.3.3.1 Treatment comparison of parasitoid abundance and taxon richness

The mean abundance and genera richness of parasitoids collected in each margin treatment was compared using the R statistical package, version 2.4.1 (R Development Core Team, 2006) to fit generalised linear mixed models with penalised quasi-likelihood (glmmPQL). A maximal initial model with a Poisson error structure, including margin width, treatment and sampling month as fixed effects, and year of sampling as a random effect, was fitted first. Non-significant terms were systematically removed in a process of model simplification using AIC values (Crawley, 2005) to determine the ‘best-fitting’ minimal model.

4.3.3.2 Testing the relationship between parasitoid abundance and taxon richness and total arthropod diversity

After pooling all samples from individual plots, and so using plots as replicates, the relationship between the taxon richness of each major arthropod group, in turn, and the total taxon richness of all other arthropod groups (excluding the group being evaluated – see Sauberer *et al.* (2004) was determined using linear model (regression) analysis using the R statistical package (version 2.4.1) (R Development Core Team, 2006). The relationship between the total abundance of parasitoid Hymenoptera and the taxon richness of other arthropods was similarly investigated.

4.3.3.3 Community structure and host relationships of parasitoid populations collected from field margin treatments

The structure of parasitoid communities collected from field margin treatments was investigated using non-metric multidimensional scaling (NMDS). NMDS is one of the most generally effective ordination methods for quantifying and interpreting community structure in ecological data and is recommended by McCune and

Grace (2002). It should be noted that NMDS differs from many other ordination techniques in that the axis numbers are arbitrary, so that the amount of variance on a given axis does not necessarily form a descending series with increasing axis number. Hence, Axis 3 may account for more variance than either Axis 1 or 2 (McCune and Grace, 2002). The analysis was performed using the PC-ORD program (McCune and Mefford, 2006). All samples collected from individual margin plots during 2004 and 2005 were pooled to create a single sample providing the most comprehensive description of parasitoid community structure in each plot. Counts of genera were log transformed $\log_{10}(n+1)$ before running an NMDS analysis in 'autopilot mode' using Sorenson Distance with a random starting configuration and 50 runs with the data. Monte Carlo tests were used to evaluate whether the extracted axes ordinations differed from random chance ($p < 0.05$), and to determine the most appropriate dimensionality (McCune and Mefford, 2006).

Information concerning the developmental biology and host range of all the parasitoid taxa collected from the field margins was obtained from published literature. Taxa were classified to guilds, as either idiobionts or koinobionts, and parasitoids of either particular taxonomic host groups (aphids, Hemiptera (other than aphids), Coleoptera, Diptera, or Lepidoptera) or of particular ecological host groups (fungal-feeders, dung-breeders, plant-mining/gall-formers, insect/spider eggs) or 'other' ecological host types. As described above, the taxa in different guild groupings were pooled across all sampling dates to create a single data set for each treatment plot. The data were analysed by ANOVA on $\log_{10}(x+1)$ data using the statistical package R, version 2.4.1 (R Core Development Team, 2006).

4.3.3.4 Indicator values

The above analyses provided useful information regarding the relationship between parasitoid communities and the experimental field margin treatments. In addition, the indicator value (IV) method of Dufrene and Legendre (1997) was used to provide additional information concerning the specific 'association' of specific genera or guild groupings with particular margin treatments. This method combines

information on the relative abundance (specificity of abundance in plots of a particular treatment) and the relative frequency of occurrence (constancy of presence in plots of a particular treatment) (Dufrene and Legendre, 1997; McCune and Grace, 2002; McGeoch *et al.*, 2002). The IV was calculated as follows:

Relative abundance (specificity measure):

a_{ijk} = the abundance of taxon j in plot i of treatment k
 n_k = the number of samples (plots) from treatment k
 g = the total number of treatments compared

First calculate mean abundance (x_{kj}) of taxon j in treatment k :

x_{kj} = the sum of a_{ijk}/n_k .

Then calculate the relative abundance (RA_{kj}) of taxon j in treatment k :

RA_{kj} = the x_{kj} /sum of x_{kj} in all treatments.

Relative Frequency (RF_{kj}) of taxon j in treatment k :

RF_{kj} = a_{ijk} (transformed to presence/absence)/ n_k

The percentage IV for taxon j in treatment k is then calculated as:

$IndVal_{kj} = 100 (RA_{kj} \times RF_{kj})$

Indicator values range from 0 to 100 and a 'perfect indicator' is one which is unique to and always occurs in that particular environment, and so has an IV of 100 (Dufrene and Legendre, 1997). Indicator values greater than 70% can be considered representative indicators for the locations in which they occur (McGeoch *et al.*, 2002). Indicator values were calculated for the individual parasitoid genera, and for parasitoid guilds (based on the host ranges and developmental types listed in Section 4.3.3.3), collected from the unfenced (grazed) paddock margins and from the fenced margin plots using the PC-ORD program. The statistical significance of the maximum IV obtained for individual genera and guilds was evaluated by the Monte Carlo method (McCune and Grace, 2002).

4.4 Results

4.4.1 Parasitoid abundance and taxon richness

Over the four sampling occasions (2 seasons $\times$ 2 years), a total of 5,466 individuals representing 117

parasitoid taxa were collected from a total sampled area of 27.36 m² of grassland margin.

Field margin width and year of sampling had no effect on either parasitoid abundance or taxon richness. Significant interaction effects between margin treatments and sample month were observed for parasitoid abundance (Table 4.1). A significant difference in total parasitoid numbers per sample between all fenced margin treatments (FO, ROT, RS) and UC margin strips was observed in June, but no such difference was found in August samples (Table 4.1; Fig. 4.1).

The taxon richness of parasitoid wasps was similarly subject to interaction effects between margin treatments and sample month (Table 4.1). The FO, ROT and RS treatments had greater taxon richness than the UC margins in June; however, there was no difference between these treatments in August. The RS treatment had greater parasitoid taxon richness than the UC margins in both seasons (Fig. 4.2).

4.4.2 The potential of parasitoids as bioindicators of other arthropod diversity

The relationship between the taxon richness of individual arthropod groups and the richness of all other taxa was significant for all groups except Diptera, although r^2 values were low (Table 4.2). However, as in previous analyses of data from the initial ten Ag-Biota monitoring sites and the 50-site survey, the genera richness and abundance of parasitic

Hymenoptera showed the strongest relationships with overall biodiversity (Table 4.2).

4.4.3 Community structure and ecology of parasitoids

The parasitic Hymenoptera collected from the field margin and grazed field have a wide range of hosts, including plant-mining insects, Lepidoptera larvae, Aphididae, Coleoptera, Diptera, other Hemiptera and mixed generalists, with probably the majority of collected taxa being parasitoids of various plant-mining insect groups and the gall-forming cecidomyiid flies (Table 4.3).

4.4.3.1 NMDS analysis of parasitoid community structure

NMDS analysis of community structure using individual parasitoid genera produced a final three-dimensional ordination with a stress of 15.05 and a final instability of 0.00001 from 172 iterations. A final stress of between 10 and 20 can correspond to a meaningful ordination, but values towards the upper end of this range need careful interpretation (Clarke, 1993). A Monte Carlo test on 50 randomised runs gave a p-value of 0.0196 for the first three axes, indicating that the final stress was unlikely to have been obtained by chance. Axes 1 and 2 accounted for 61.9% and 10.1% of the variance (approximately 72% in combination), respectively, and show a clear separation of parasitoid community structure in UC margins from all fenced margins. (Fig. 4.3) This is most

Table 4.1. Results¹ from analysis of the effects of field margin treatments on the abundance and taxon richness of parasitoid Hymenoptera using generalised linear mixed models (glmmPQL) with Poisson error structure.

	Minimal model	Parameter	Parameter estimate	Std error	df	t	p-value
Parasitoid abundance	Treatment × month	UC	3.911	0.09	135	42.33	0.000
		FO x month	0.541	0.26	135	2.096	0.038
		ROT x month	0.652	0.25	135	2.607	0.010
		RS x month	0.527	0.25	135	2.102	0.038
Taxon richness	Treatment × month	UC	2.856	0.07	135	41.38	0.000
		FO x month	0.345	0.16	135	2.225	0.028
		ROT x month	0.329	0.15	135	2.191	0.030
		RS x month	0.200	0.15	135	1.326	0.187

¹An initial maximal model included margin width, treatment and sampling month as fixed effects, and year of sampling as a random effect. Margin width and sample year effects were not significant. Only details for the final minimal model are shown. UC, unfenced control; FO, fenced only; ROT, rotavated and fenced; RS, rotavated, reseeded and fenced. x signifies interaction.

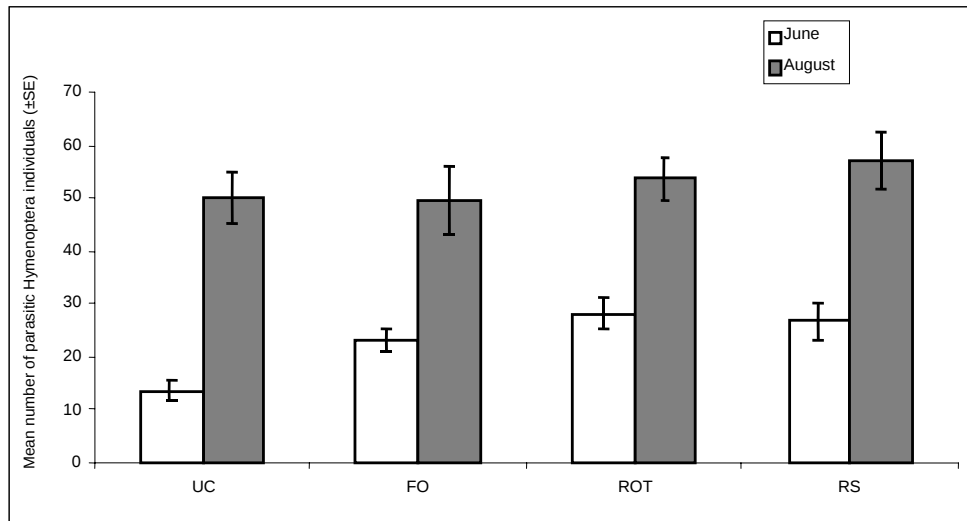


Figure 4.1. The response of parasitic Hymenoptera abundance to fenced field margin treatments and sampling month at Johnstown Castle, Co. Wexford. UC, unfenced control; FO, fenced only, ROT, rotavated and fenced; RS, rotavated, reseeded and fenced. The bars represent standard errors of the means.

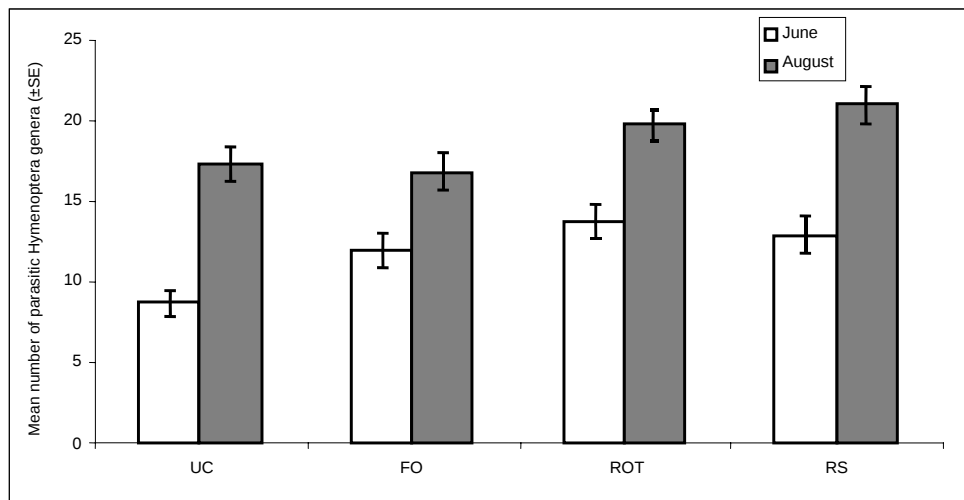


Figure 4.2. The response of parasitic Hymenoptera genera richness to fenced field margin treatments and sampling month at Johnstown Castle, Co. Wexford. UC, unfenced control; FO, fenced only, ROT, rotavated and fenced; RS, rotavated, reseeded and fenced. The bars represent standard errors of the means.

apparent on Axis 1 where there is no overlap between these treatments. The majority of parasitoid genera are clearly associated with the fenced margins; however, some taxa, particularly the genera *Trioxys*, *Chasmodon*, *Polynema*, and *Phaenocarpa*, were found to be relatively more characteristic of the UC (grazed) paddock margins. There was no evidence of divergence in parasitoid community structures in the different fenced margin treatments.

4.4.3.2 Occurrence of ecological parasitoid groups

ANOVA was used to assess field margin treatment effects on the incidence of different parasitoid groups, including the incidence of idiobiont and koinobiont species, and groups with specific host types.

(a) Abundance of different ecological groups: The abundance of parasitoids with idiobiont development, which kill or immediately paralyse their host to prevent

Table 4.2. Summary of regression analyses investigating the taxon richness of an individual group and the overall richness of all other groups from the field margin experiment.

Major group (level of taxonomic resolution)	r^2	p-value*
Coleoptera (species)	0.210	<0.001
Diptera (family)	0.006	0.369
Hymenoptera (family)	0.148	<0.001
Hymenoptera (genus)	0.244	<0.001
Hymenoptera (abundance)	0.287	<0.001
Hemiptera (species)	0.079	<0.001
Araneae (species)	0.049	0.008

*Bonferroni-adjusted critical p-value = $0.05/7 = 0.0074$.

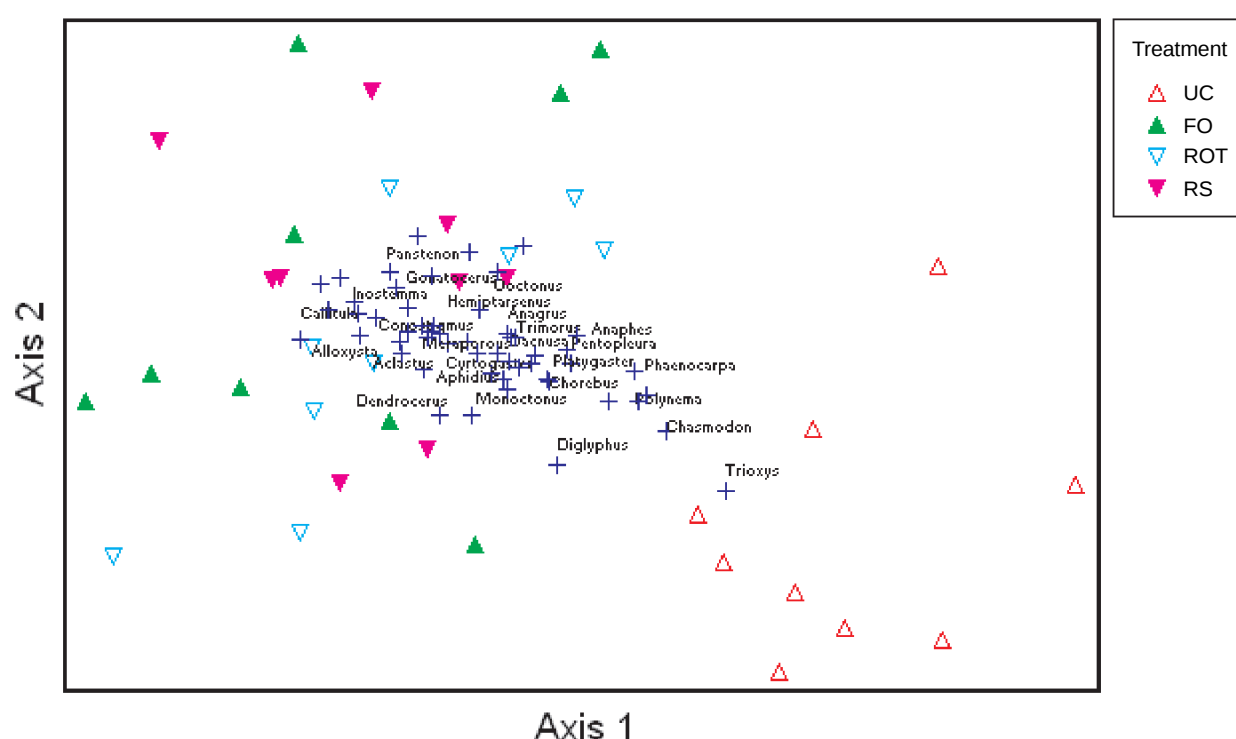


Figure 4.3. NMDS ordination for parasitoid Hymenoptera collected from the plots of the field margin experiment at Johnstown Castle. The blue crosses represent the ordination of individual parasitoid genera relative to that of treatment plots. UC, unfenced control; FO, fenced only, ROT, rotavated and fenced; RS, reseeded.

its further development, was significantly different between margin treatments (Table 4.4), with smaller numbers obtained from UC (grazed) margins compared with all fenced treatments (Fig. 4.4). The abundance of koinobiont species was similarly less in the UC plots (Fig. 4.5) but, overall, the difference between treatments was barely significant at the $p = 0.05$ level (Table 4.4).

The total abundance of parasitoids of gall-forming hosts, and of larval Lepidoptera also showed significant differences ($p < 0.05$) between the paddock margin treatments (Table 4.4), with all the fenced margin treatments having greater abundance of these parasitoid guilds than the UC (grazed) margins (Figs 4.6 and 4.7). The abundance of parasitoids of Diptera larvae and of fungal-feeding insect larvae was similarly

Table 4.3. Genera of parasitic Hymenoptera collected from field margins and their host ranges obtained from the literature.

Family	Genus	Host range	Reference
Aphelinidae	<i>Aphelinus</i>	Aphididae (Hemiptera)	Woolley, 1997
Braconidae	<i>Aphidius</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Binodoxys</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Diaeretiella</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Diaeretiellus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Ephedrus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Monoctonus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Trioxys</i> *	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Meteorus</i>	Coleoptera and Lepidoptera larvae	Shaw, 1997c
Braconidae	<i>Centistes</i>	Coleoptera larvae	Shaw, 1997b
Braconidae	<i>Blacus</i>	Coleoptera larvae	Sharkey, 1997
Braconidae	<i>Phaenocarpa</i> ¹	Dung or fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Pentapleura</i>	Dung-breeding Diptera	Wharton, 1997a
Braconidae	<i>Aspilota</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Dinotrema</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Orthostigma</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Chasmodon</i> ¹	Leaf-mining dipteran larvae	Umoru, 1997
Braconidae	<i>Chelonus</i>	Lepidoptera larvae	Shaw, 1997a
Braconidae	<i>Cotesia</i>	Lepidoptera larvae	Whitfield, 1997
Braconidae	<i>Macrocentrus</i>	Lepidoptera larvae	Wharton, 1997b
Braconidae	<i>Microplitis</i>	Lepidoptera larvae	Whitfield, 1997
Braconidae	<i>Alloea</i>	Lonchopteridae (fungal-feeding Diptera larvae)	Wharton, 1997a
Braconidae	<i>Peristenus</i>	Miridae (Hemiptera) nymphs and adult	Shaw, 1997b
Braconidae	<i>Opius</i>	Plant-mining Curculionidae (Coleoptera) and Cecidomyiidae (Diptera) larvae	Wharton, 1997c
Braconidae	<i>Alysia</i>	Plant-mining Curculionidae eggs (Coleoptera)	Wharton, 1997a
Braconidae	<i>Chorebus</i>	Plant-mining Diptera larvae	Wharton, 1997a
Braconidae	<i>Dacnusa</i>	Plant-mining Diptera larvae	Wharton, 1997a
Braconidae	<i>Dapsilarthra</i>	Plant-mining Diptera larvae	Wharton, 1997a
Ceraphronidae	<i>Aphanogmus</i>	Cecidomyiidae	Alekseev, 1987
Ceraphronidae	<i>Ceraphron</i>	Gall-forming Cecidomyiidae larvae (Diptera)	Alekseev, 1987
Diapriidae	<i>Aclista</i>	Fungal-feeding Diptera larvae	Nixon, 1957
Diapriidae	<i>Basalys</i>	Fungal-feeding Diptera larvae	Nixon, 1980
Diapriidae	<i>Pamis</i>	Fungal-feeding Diptera larvae	Nixon, 1957
Diapriidae	<i>Trichopria</i>	Plant-mining Curculionidae (Coleoptera) and Cecidomyiidae (Diptera) larvae	Nixon, 1980
Dryinidae	<i>Anteon</i>	Homoptera (Hemiptera) nymphs	Ponomarenko, 1987
Dryinidae	<i>Gonatopus</i>	Homoptera (Hemiptera) nymphs	Ponomarenko, 1987
Encyrtidae	<i>Copidosma</i>	Lepidoptera larvae	Noyes <i>et al.</i> , 1997
Encyrtidae	<i>Anagyrus</i>	Pseudococcidae (Hemiptera)	Noyes <i>et al.</i> , 1997
Encyrtidae	<i>Rhopus</i>	Pseudococcidae (Hemiptera)	Noyes <i>et al.</i> , 1997
Eulophidae	<i>Asecodes</i>	Coleoptera larvae	Schauff <i>et al.</i> , 1997
Eulophidae	<i>Aprostocetus</i>	Gall-forming Cecidomyiidae larvae	Graham, 1987; Schauff <i>et al.</i> , 1997
Eulophidae	<i>Omphale</i>	Gall-forming Cecidomyiidae larvae	Schauff <i>et al.</i> , 1997

Table 4.3 *contd.*

Family	Genus	Host range	Reference
Eulophidae	<i>Chrysocharis</i>	Leaf-mining insect larvae	Schauff <i>et al.</i> , 1997
Eulophidae	<i>Hemiptarsenus</i>	Leaf-mining insect larvae	Askew, 1968b
Eulophidae	<i>Diglyphus</i> ¹	Plant-mining Diptera larvae	Askew, 1968b
Eulophidae	<i>Pediobius</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera)	Schauff <i>et al.</i> , 1997
Eulophidae	<i>Closterocerus</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera, Homoptera)	Schauff <i>et al.</i> , 1997
Eurytomidae	<i>Tetramesa</i>	Gall-forming insect larvae	Di Giulio, 1997
Eurytomidae	<i>Eurytoma</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera, Diptera, Homoptera)	Di Giulio, 1997
Figitidae	<i>Melanips</i>	Diptera larvae	Fergusson, 1986
Figitidae	<i>Sarothrus</i>	Diptera larvae	Fergusson, 1986
Figitidae	<i>Kleidotoma</i>	Dung or fungal-feeding Diptera	Quinlan, 1978; Wharton, 1997a
Figitidae	<i>Phaenoglyphis</i>	Hyperparasitoid via Aphidiinae	Fergusson, 1986
Figitidae	<i>Alloxysta</i>	Hyperparasitoids via Aphidiinae	Fergusson, 1986
Figitidae	<i>Rhoptromeris</i>	Leaf-mining insect larvae	Quinlan, 1978
Figitidae	<i>Anacharis</i>	Neuroptera larvae	Fergusson, 1986
Ichneumonidae	<i>Phygadeuon</i>	Diptera pupae	Townes, 1970a
Ichneumonidae	<i>Mesochorus</i>	Hyperparasites of Braconidae and Ichneumonidae	Townes, 1971
Ichneumonidae	<i>Amblyteles</i>	Lepidoptera larvae	Perkins and Eady, 1960
Ichneumonidae	<i>Diadegma</i>	Lepidoptera larvae	Azidah <i>et al.</i> , 2000
Ichneumonidae	<i>Lissonota</i>	Lepidoptera larvae	Townes, 1970b
Ichneumonidae	<i>Epitomus</i>	Lepidoptera pupae	Perkins and Eady, 1960
Ichneumonidae	<i>Limerodops</i>	Lepidoptera pupae	Perkins and Eady, 1960
Ichneumonidae	<i>Stenomacrus</i>	Mycetophilidae larvae (fungal-feeding Diptera)	Townes, 1971
Ichneumonidae	<i>Promethes</i>	Syrphidae (Diptera) larvae	Fitton and Rotheray, 1982
Ichneumonidae	<i>Perilissus</i>	Tenthredinidae (Hymenoptera) larvae	Townes, 1970b
Ichneumonidae	<i>Endasys</i>	Tenthredinidae (Hymenoptera) pupae	Townes, 1970a
Ichneumonidae	<i>Aclastus</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera)	Townes, 1970a
Ichneumonidae	<i>Gelis</i>	Various cocoons (e.g. Lepidoptera, Hymenoptera, spider cocoons)	Schwartz and Shaw, 1999
Megaspilidae	<i>Dendrocerus</i>	Hyperparasites via Aphidoidea	Fergusson, 1980
Megaspilidae	<i>Conostigmus</i>	Syrphidae (Diptera) larvae	Alekseev, 1987
Megaspilidae	<i>Lagynodes</i>	Unknown	Alekseev, 1987
Mymaridae	<i>Ooconus</i>	Cercopidae, Cicadellidae eggs (Hemiptera)	Huber, 1997
Mymaridae	<i>Gonatocerus</i>	Cicadellidae, Membracidae eggs (Hemiptera)	Huber, 1997
Mymaridae	<i>Polynema</i> ¹	Cicadellidae, Miridae eggs (Hemiptera)	Huber, 1997
Mymaridae	<i>Clereuchus</i>	Curculionidae eggs	Huber, 1997
Mymaridae	<i>Mymar</i>	Delphacidae, Cicadellidae eggs	Huber, 1997
Mymaridae	<i>Anagrus</i>	Hemiptera eggs	Huber, 1997
Mymaridae	<i>Alaptus</i>	Psocoptera eggs	Huber, 1997
Mymaridae	<i>Litus</i>	Staphylinidae (Coleoptera) eggs	Huber, 1997
Mymaridae	<i>Anaphes</i>	Various eggs	Huber, 1997
Platygastridae	<i>Synopeas</i>	Gall-forming Cecidomyiidae eggs or larvae	Kozlov, 1987
Platygastridae	<i>Leptacis</i>	Gall-forming Cecidomyiidae eggs/larvae	Kozlov, 1987
Platygastridae	<i>Platygaster</i>	Gall-forming Cecidomyiidae eggs/larvae	Kozlov, 1987
Platygastridae	<i>Inostemma</i>	Gall-forming Cecidomyiidae larvae	Kozlov, 1987

Table 4.3 contd.

Family	Genus	Host range	Reference
Proctotrupidae	<i>Codrus</i>	Coleoptera larvae in soil litter	Kozlov, 1987
Proctotrupidae	<i>Phaenoserphus</i>	Coleoptera larvae in soil litter	Kozlov, 1987
Pteromalidae	<i>Meraporus</i>	Curculionidae; Cecidomyiidae	Bouček and Rasplus, 1991
Pteromalidae	<i>Trichomalus</i>	Curculionidae; plant-mining Diptera	Bouček and Rasplus, 1991
Pteromalidae	<i>Rhynocoelia</i>	Delphacidae and Cicadellidae eggs (Hemiptera)	Bouček and Rasplus, 1991
Pteromalidae	<i>Spalangia</i>	Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Gastrancistrus</i>	Gall-forming Cecidomyiidae larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Macroglenes</i>	Gall-forming Cecidomyiidae larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Spaniopus</i>	Gall-forming Cecidomyiidae larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Semiotellus</i>	Gall-forming Cecidomyiidae pupae	Bouček and Rasplus, 1991
Pteromalidae	<i>Asaphes</i>	Hyperparasites via Aphidiinae	Bouček and Rasplus, 1991
Pteromalidae	<i>Thinodytes</i>	Leaf-mining Agromyzidae larvae (Diptera)	Bouček and Rasplus, 1991
Pteromalidae	<i>Spintherus</i>	Plant-mining Apionidae larvae (Coleoptera)	Bouček and Rasplus, 1991
Pteromalidae	<i>Callitula</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Chlorocytus</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Cyrtogaster</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Halticoptera</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Homoporus</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Merismus</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Miscogaster</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Stenomalina</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Mesopolobus</i>	Plant-mining insect larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Seladerma</i>	Plant-mining insect larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Panstenon</i>	Predators Delphacidae (Hemiptera) eggs	Bouček and Rasplus, 1991
Pteromalidae	<i>Trichomalopsis</i>	Various pupae (e.g. Lepidoptera, Coleoptera, Diptera, Hymenoptera)	Bouček and Rasplus, 1991
Scelionidae	<i>Baeus</i>	Insect and spider eggs	Polaszek and Notton, 2003
Scelionidae	<i>Telenomus</i>	Insect and spider eggs	Polaszek and Notton, 2003
Scelionidae	<i>Trimorus</i>	Insect and spider eggs	Polaszek and Notton, 2003
Tetracampidae	<i>Epiclerus</i>	Leaf-mining insect larvae	Bouček and Heydon, 1997

¹Taxa associated with unfenced control (grazed) paddock margins in NMDS ordination (see Fig. 4.3).

less than in all fenced margin treatments; however, these treatment differences were not statistically significant at $p = 0.0689$ and 0.0829 , respectively. There was no difference in the abundance of plant-mining insect parasitoids between the margin treatments (Table 4.4).

(b) Taxon richness in different ecological groups:

The taxon richness of idiobionts, of parasitoids of Lepidoptera, Hemiptera (other than aphids), of insect and spider eggs and of gall-forming insect larvae showed significant differences between grazed paddock margin treatments (Table 4.5).

All of these groups showed least taxon richness in the fenced margin treatments compared with all UC (grazed) margins, which were generally not significantly (Figs. 4.8–4.12). However, the numbers of insect and spider egg parasitoid taxa were similar in the UC (grazed) margins and margins of the FO treatment, with significantly greater numbers of such egg parasitoid taxa collected from the ROT and the RS treatments, which showed similar taxon richness for this parasitoid group (Fig. 4.9).

The relative absence of parasitoid taxa in the UC margins was most marked for parasitoids of

Table 4.4. Summary of ANOVA ($\log_{10}(x+1)$ transformed data) comparing the abundance (number of individuals) of parasitoid guilds collected from the field margin experiment at Teagasc, Johnstown Castle, Co. Wexford. Significant ($p < 0.05$) results are emboldened.

Group	df	F	p
Idiobionts	3,6	5.886	0.031
Koinobionts	3,6	4.490	0.056
Hyperparasitoids	3,6	3.217	0.104
Parasitoids of (taxonomic host groups):			
Hemiptera: Aphididae	3,6	1.613	0.283
Hemiptera (other than aphids)	3,6	2.080	0.204
Coleoptera larvae	3,6	0.840	0.520
Diptera larvae	3,6	4.038	0.069
Lepidoptera larvae	3,6	6.891	0.023
Parasitoids of (ecological host groups):			
Dung-feeding insect larvae	3,6	1.361	0.341
Fungal-feeding insect larvae	3,6	3.652	0.083
Plant-mining insect larvae	3,6	0.394	0.762
Gall-forming insect larvae	3,6	16.960	0.003
Insect and spider eggs	3,6	2.231	0.185

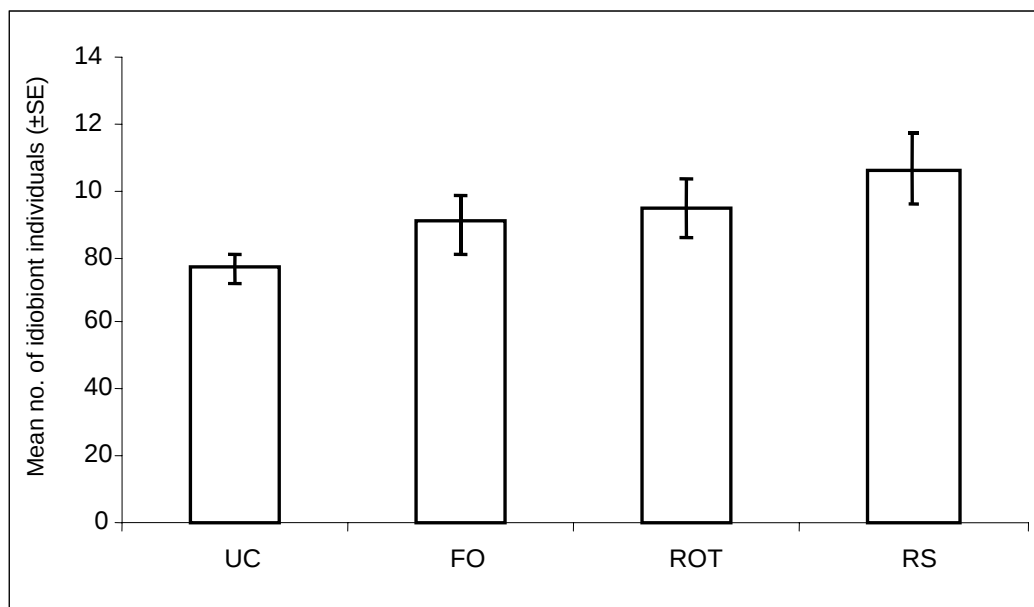


Figure 4.4. Estimated mean total abundance of idiobiont parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

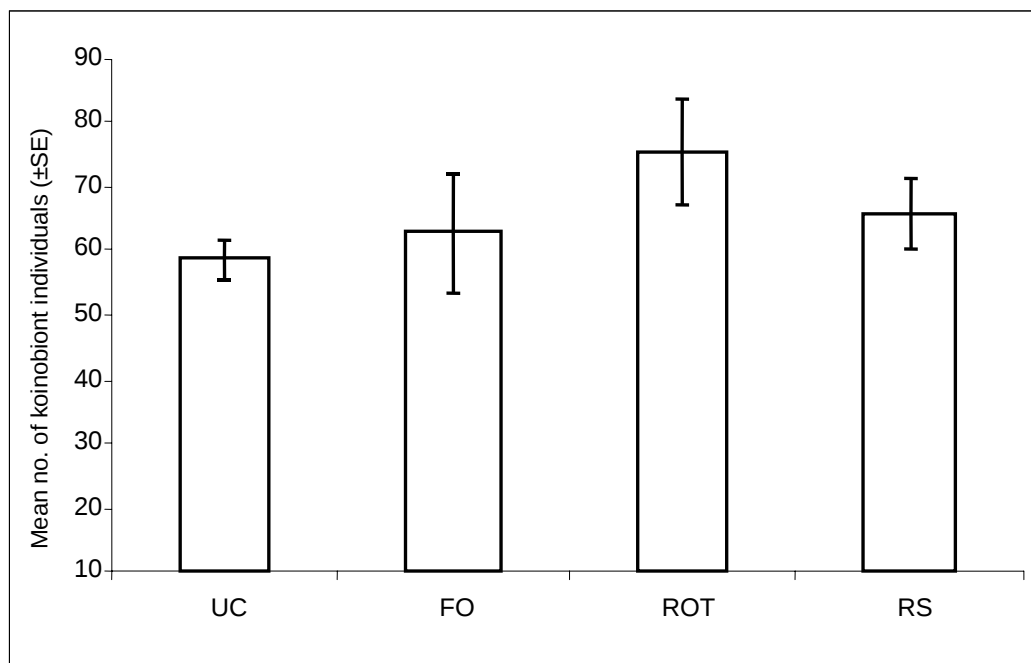


Figure 4.5. Estimated mean total abundance of koinobiont parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

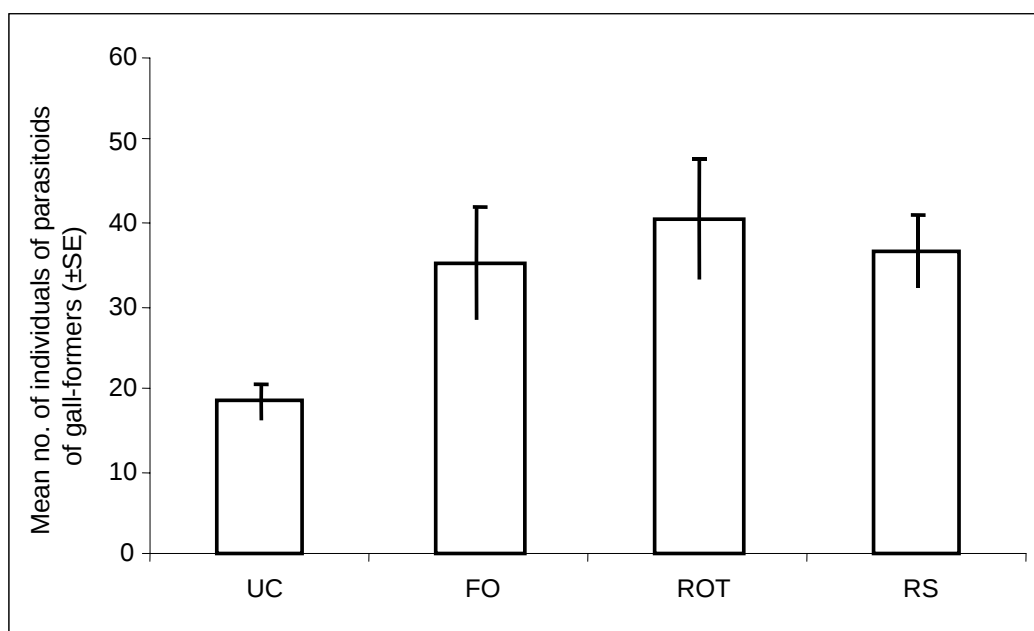


Figure 4.6. Estimated mean total abundance of gall-forming insect larval parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

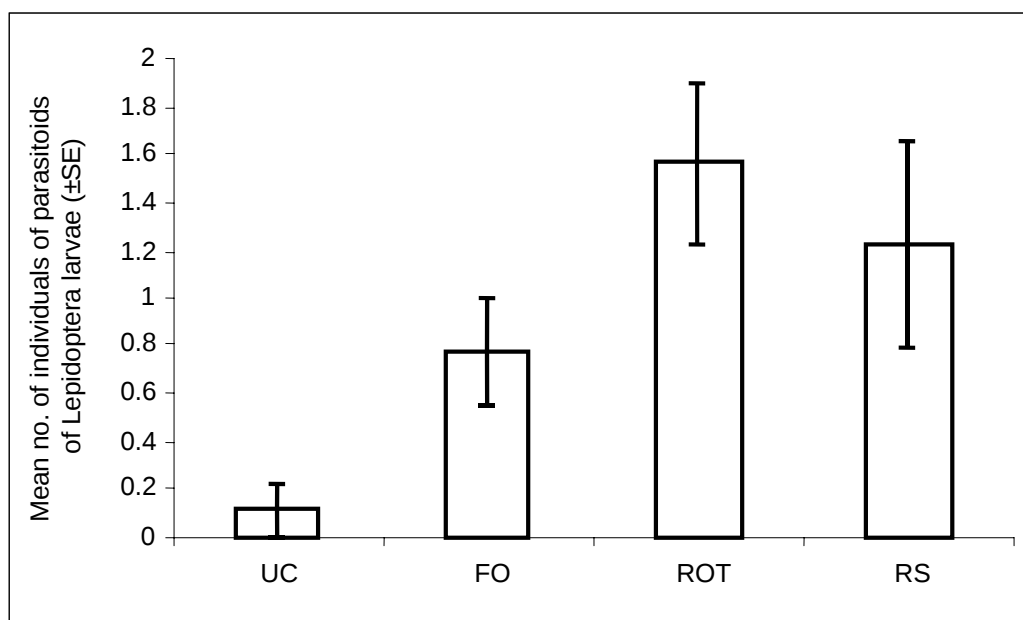


Figure 4.7. Estimated mean total abundance of lepidopterous larval parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

Table 4.5. Summary of ANOVA ($\log_{10}(x+1)$ transformed data) comparing the taxon richness of parasitoid groups with different biologies and host ranges in the field margin experiment at Teagasc, Johnstown Castle, Co. Wexford. Significant ($p < 0.05$) results are shown in bold.

Parasitoid group	df	F	p
Idiobiont	3,6	34.571	<0.001
Koinobiont	3,6	3.279	0.102
Parasitoids of (taxonomic host groups):			
Hemiptera: Aphididae	3,6	3.259	0.102
Hemiptera (other than aphids)	3,6	13.391	0.005
Coleoptera larvae	3,6	0.840	0.520
Diptera larvae	3,6	0.359	0.785
Lepidoptera larvae	3,6	7.000	0.022
Parasitoids of (ecological host groups):			
Dung-feeding insect larvae	3,6	1.361	0.341
Fungal-feeding insect larvae	3,6	2.407	0.166
Plant-mining insect larvae	3,6	3.267	0.101
Gall-forming insect larvae	3,6	7.724	0.018
Insect and spider eggs	3,6	16.337	0.003

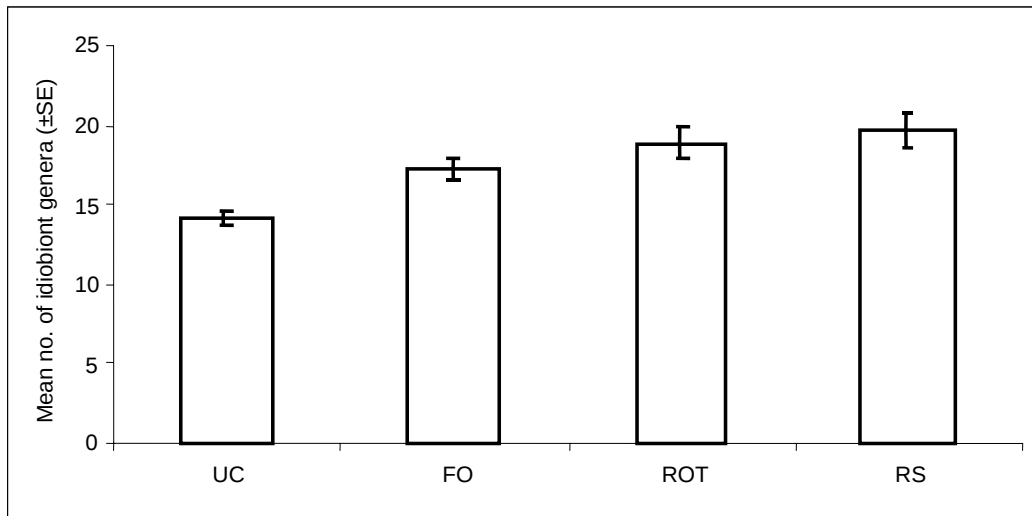


Figure 4.8. Estimated mean taxon richness of idiobiont parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

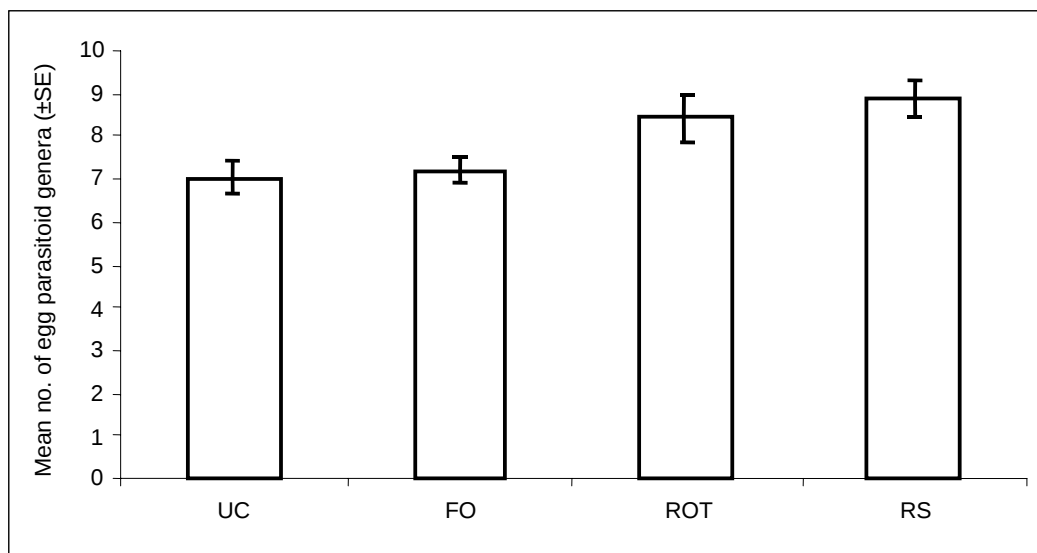


Figure 4.9. Estimated mean taxon richness of insect and spider egg parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT,) and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

lepidopterous larvae and of Hemiptera – other than aphids (Figs 4.11, 4.12). In the case of lepidopteran parasitoids, taxon richness in the FO margins was also less than in the more heavily manipulated ROT and RS margins.

4.4.4 The indicator value of parasitoid taxa

Indicator values were calculated for all the parasitoid genera collected, contrasting populations from the UC (grazed) paddock margins with populations collected from all fenced treatment plots. Taxa with significant

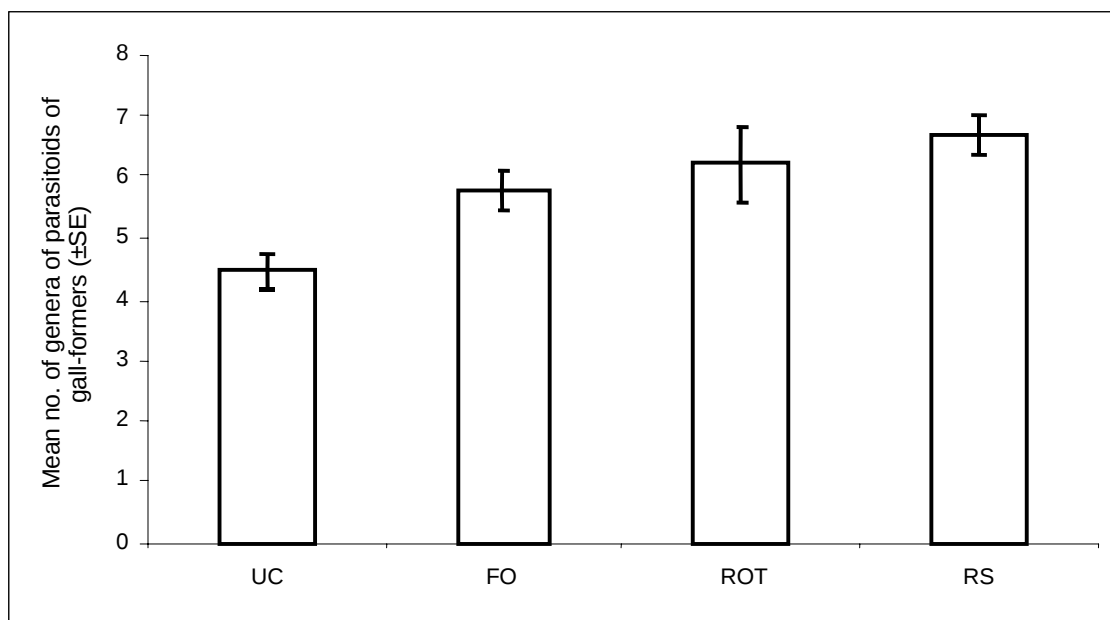


Figure 4.10. Estimated mean taxon richness of gall-forming larval insect parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

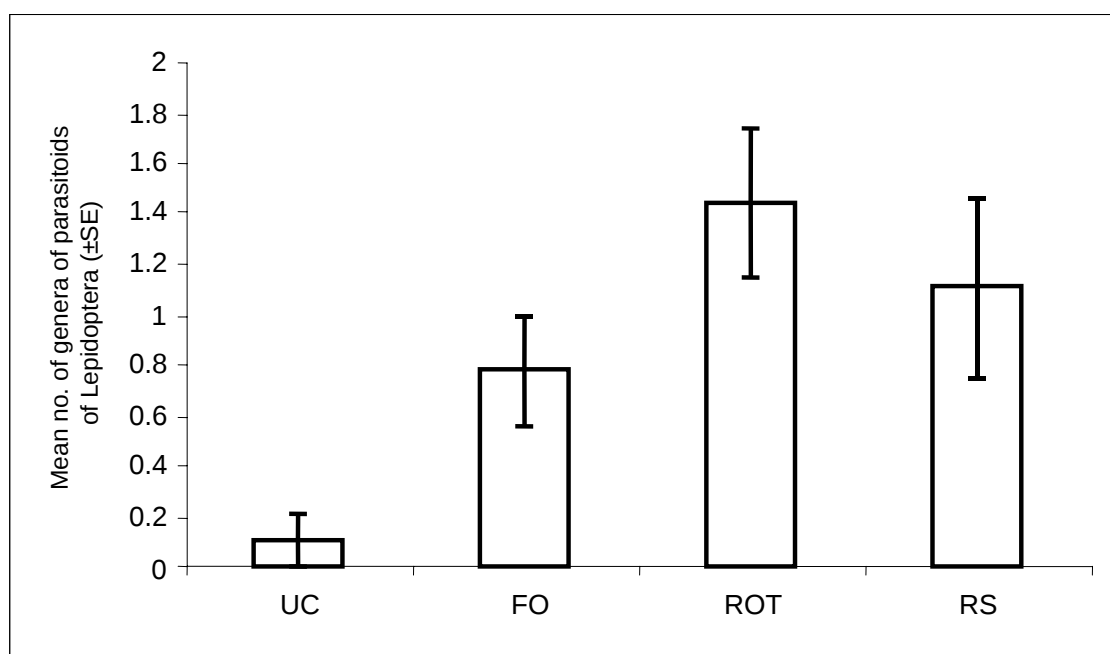


Figure 4.11. Estimated mean taxon richness of Lepidoptera larval parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

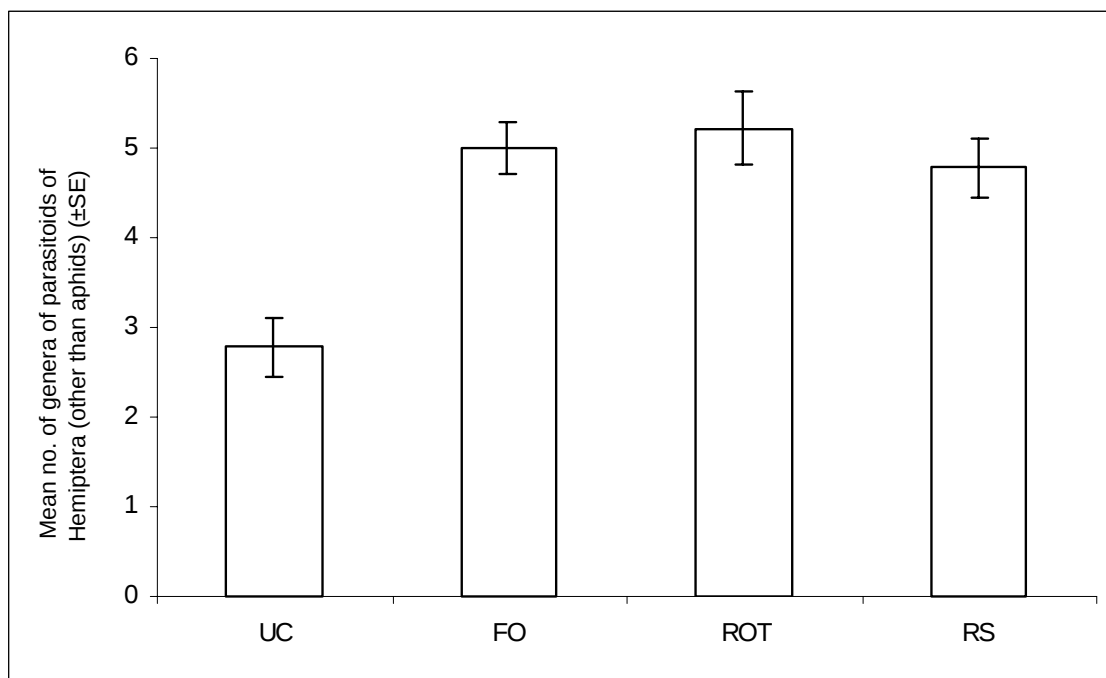


Figure 4.12. Estimated mean taxon richness of Hemiptera (other than aphids) parasitoids in suction samples collected from unfenced (grazed) control margins (UC), fenced only (FO), rotavated and fenced (ROT), and rotavated, reseeded and fenced (RS) treatments in the field margin experiment at Teagasc, Johnstown Castle. The bars represent standard errors of the means.

($p = 0.05$) IVs greater than 70% can be considered good indicators of the habitats in which they are found (McGeoch *et al.*, 2002) (Table 4.6). Only genera with significant ($p = 0.05$) IVs are shown in this table and taxa with IVs $\geq 70\%$ (emboldened) can be considered representative of the locations they are associated with (McGeoch *et al.*, 2002). Seven genera met this criterion: *Polynema* and *Chasmodon* were associated with the UC (grazed) paddock margin, and *Inostemma*, *Panstenon*, *Anagyrus*, *Gonatocerus* and *Mesopolobus* with fenced margins (Table 4.6). *Polynema* is a parasitoid of the eggs of hemipteran leaf-hoppers and mirid bugs, and *Chasmodon* parasitises plant-mining dipteran larvae. *Inostemma* parasitises Cecidomyiidae eggs or larvae and *Panstenon* is predatory on plant-hopper (delphacid) eggs. *Anagyrus* and *Gonatocerus* are known to parasitise the eggs of pseudococcids and hemipteran cicadellids, respectively, while *Mesopolobus* is a parasitoid of plant-mining Diptera. As it is probably unlikely that there would be significant numbers of pseudococcid hosts in Irish grassland, the known host range of *Anagyrus* may well be deficient.

4.4.5 The indicator value of parasitoid guilds

An IV analysis was done to contrast parasitoid guild groupings in UC (grazed), and all fenced margins, combining data for 2004 and 2005. As with the NMDS analysis above, all samples collected from individual margin plots during 2004 and 2005 were pooled to create a single data set providing the most comprehensive description of parasitoid community structure at the site. Although no guild grouping had a sufficiently large IV ($\geq 70\%$) to be considered a good indicator of either margin type, parasitoids of Lepidoptera, Diptera, gall-formers, and fungal-feeders all had significant IVs ($p = 0.05$) and idiobionts a close to significant IV ($p = 0.051$) (Table 4.7). Only the parasitoids of dipterous larvae were more strongly associated with UC (grazed) margins; all other groups with significant IVs were more strongly associated with fenced conservation margins (Table 4.7).

When IVs were calculated to contrast the incidence of parasitoid guilds in UC (grazed) paddock margins with margins of the other separate conservation treatments, stronger treatment associations tended to be found.

Table 4.6. Indicator values (IVs) for parasitoid genera collected in 2004 and 2005 (pooled data) from unfenced (grazed) and fenced margins in the field margin experiment at Teagasc, Johnstown Castle (pooling data for all fenced treatments). Genera with IVs greater than 70% are shown in bold.

Genus	Known host groups	Associated with	IV	p-value
<i>Aclastus</i>	Various (e.g. Lepidoptera, Hymenoptera, Diptera)	Fenced margins	66.6	0.0134
<i>Anagyrus</i>	Hemiptera: Pseudococcidae	Fenced margins	70.4	0.0012
<i>Aphanogmus</i>	Diptera: Cecidomyiidae larvae	Fenced margins	67.4	0.0094
<i>Baeus</i>	Insect and spider eggs	Unfenced margins (UC)	40.7	0.0326
<i>Basalys</i>	Fungal-feeding dipteran larvae	Unfenced margins (UC)	55.3	0.202
<i>Callitula</i>	Plant-mining dipteran larvae	Fenced margins	68.7	0.0116
<i>Chasmodon</i>	Plant-mining dipteran larvae	Unfenced margins (UC)	77.5	0.0002
<i>Chorebus</i>	Plant-mining dipteran larvae	Unfenced margins (UC)	66.9	0.0056
<i>Diglyphus</i>	Plant-mining dipteran larvae	Unfenced margins (UC)	61.8	0.0062
<i>Gonatocerus</i>	Hemiptera: Cicadellidae, Membricidae	Fenced margins	70.9	0.0016
<i>Hemiptarsenus</i>	Plant-mining dipteran larvae	Fenced margins	64.4	0.0126
<i>Inostemma</i>	Diptera: Cecidomyiidae eggs/larvae	Fenced margins	92.2	0.0002
<i>Mesopolobus</i>	Plant-mining dipteran larvae	Fenced margins	70.4	0.0010
<i>Ooetonus</i>	Cercopidae, Cicadellidae (Hemiptera) eggs	Fenced margins	56.7	0.0196
<i>Opius</i>	Diptera	Unfenced margins (UC)	62.3	0.0492
<i>Panstenon</i>	Hemiptera: Delphacidae eggs	Fenced margins	93.7	0.0002
<i>Pediobius</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera)	Fenced margins	40.7	0.0574
<i>Phaenocarpa</i>	Dung-feeding Diptera larvae	Unfenced margins (UC)	59.2	0.0174
<i>Polynema</i>	Hemiptera: Cicadellidae eggs	Unfenced margins (UC)	86.5	0.0002
<i>Rhopus</i>	Hemiptera: Pseudococcidae	Fenced margins	44.4	0.0394
<i>Spaniopus</i>	Diptera: Cecidomyiidae	Fenced margins	58.6	0.0128
<i>Telenomus</i>	Insect and spider eggs	Fenced margins	63.0	0.0040
<i>Trichopria</i>	Diptera	Unfenced margins (UC)	67.3	0.0094
<i>Trichomalopsis</i>	Various pupae	Fenced margins	55.6	0.0092
<i>Trioxys</i>	Hemiptera: Aphididae	Unfenced margins (UC)	39.0	0.0184

Table 4.7. Indicator values (IVs) for parasitoid guilds (bio-ecological groups) collected from unfenced (grazed) and fenced margins in the field margin experiment at Teagasc, Johnstown Castle (pooling data for all fenced treatments and combining data from sampling years).

Group	Associated with	IV	p-value
Idiobionts	Fenced margins	51.1	0.051
Parasitoids of Diptera larvae	Unfenced margins (UC)	58.0	0.0186
Parasitoids of Lepidoptera larvae	Fenced margins	66.5	0.0026
Parasitoids of gall-formers	Fenced margins	54.9	0.0028
Parasitoids of fungal-feeders	Fenced margins	56.4	0.0026

IVs contrasting parasitoid guild incidence in UC and ROT margins are shown in Table 4.8. Parasitoids of Lepidoptera were found to be excellent indicators of the ROT margins (IV = 81.6%) compared with UC

paddock margins. Other groups show associations similar to those observed when UC (grazed) margins were compared with pooled fenced margins (see Tables 4.7 and 4.8).

Table 4.8. Indicator values (IV) for parasitoid guilds (bio-ecological groups) collected from unfenced (grazed) control (UC) and rotavated and fenced (ROT) margins in the field margin experiment at Johnstown Castle, Co. Wexford (combining data from sampling years). Genera with IVs greater than 70% are shown in bold.

Group	Associated with	IV	p-value
Idiobionts	ROT	52.3	0.0006
Parasitoids of Diptera larvae	UC	62.1	0.0136
Parasitoids of Lepidoptera larvae	ROT	81.6	0.0008
Parasitoids of gall-formers	ROT	55.4	0.0028
Parasitoids of fungal-feeders	ROT	56.3	0.0054

4.5 Discussion

Many studies have shown that conservation field margins in arable environments can enhance arthropod diversity and abundance (e.g. Meek *et al.*, 2002; Asteraki *et al.*, 2004). However, much less is known about the potential of conservation margins to enhance arthropod biodiversity within a grassland context. Increasing the structural complexity and botanical diversity of vegetation may be expected to increase the abundance and diversity of potential hosts of parasitoid species (Murphy and LaSalle, 1999). Both vegetation structure and plant species richness have been found to influence the abundance, species richness and community structure of parasitoid Hymenoptera (Chay-Hernandez *et al.*, 2006; Fraser *et al.*, 2007). In the current study of the Teagasc grassland margin experiment, further evidence was found to support the hypothesis that both the abundance and diversity of parasitoid wasps are more strongly related to the wider diversity of other arthropod taxa than any other group. The study also showed that more parasitoid genera and individuals were found in samples from the fenced margin treatments compared with the normal grazed paddock margins. In both years, this faunal difference was particularly evident in June samples, but became less obvious later in the season in August samples. Purvis and Curry (1981) observed a similar diminution of previous early summer differences in the fauna of different grassland husbandry systems, and attributed this seasonal effect to an overriding influence of hotter and drier climatic conditions as the summer advanced.

Of the parasitoid guilds that showed significant differences between the grazed and the fenced margin

treatments, the parasitoids of Lepidoptera larvae, Hemiptera (other than aphids), gall-forming Diptera larvae and parasitoids of the eggs of insects and spiders were all significantly more taxon-rich in the fenced margins in which botanical diversity was significantly greater compared to the unfenced margins of the normally grazed paddock (Sheridan *et al.*, 2008). Parasitoids of gall-forming Diptera and of lepidopterous larvae were also more abundant in terms of numbers of individuals in these fenced conservation margins.

Because of the specialised biology of parasitoid species, changes in their diversity and abundance can provide a unique insight into wider ecological changes in arthropod communities. Parasitoid incidence and species richness on herbivorous hosts is known to be greater in less mobile groups, but the numbers and incidence of parasitoid species tends to also decrease with increasing host concealment (Hawkins and Lawton, 1987) so that, for example, gall-formers are likely to have fewer parasitoids than leaf-miners, which in turn are less likely to be attacked than aphids. It is therefore telling that many of the parasitoid taxa that were found to be most obviously more abundant and diverse in the field margin treatments were in fact taxa that parasitise well-concealed larval hosts such as cecidomyiid gall midges, plant-mining larvae and insect and spider eggs. One of the most distinctive indicator groups for the field margins was the guild of lepidopteran larval parasitoids. Virtually no adult Lepidoptera were collected from any of the experimental margins during the current study. However, it is very likely that the majority of lepidopterous larval parasitoids observed in the

conservation margins are parasites of well-concealed 'micro-lepidopteran' larvae of the Family Pyralidae, including *Crambus*, *Agriphila* and *Chrysoteuchia* spp., which mine in grasses, and of other herbaceous plant species in mainly non-agricultural contexts (Goater, 1986). Observation of their parasitoids is probably the only feasible way that the presence of such cryptic biodiversity within grassland plants can be measured, short of the extremely laborious process of dissecting plants.

The idiobiont/koinobiont dichotomy can provide strong indications about the biological attributes of parasitoids associated with different types of host, and is a useful way to interpret and better understand patterns in host-parasitoid interactions (Gauld and Bolton, 1988). Idiobionts, which prevent the further development of their host upon parasitisation, tend to be more generalist parasitoids (Askew and Shaw, 1986) and dominate in concealed, plant-mining herbivore communities (Hawkins, 1994). They also tend to have longer adult lifespans (Gauld and Bolton, 1988) compared with koinobionts. Idiobionts are therefore more dependent on floral resources than koinobionts to sustain their adult life (Emden, 1962; Foster and Ruesink, 1984; Jervis *et al.*, 1993). The idiobiont developmental strategy seems to be well adapted to the parasitism of hosts that are not likely to be taken by other predators following parasitism (Hawkins, 1994). The incidence of idiobiont species is known to increase with increasing plant structural diversity. In the current study, both the abundance and taxon richness of idiobionts were substantially greater in the structurally more diverse fenced margin treatments compared to the unfenced paddock margins.

In contrast, it is likely that koinobiont parasitoids have had to become much more closely adapted to their hosts, in order to ensure their continued development in the host following parasitism when the latter occurs in an exposed habitat, and would otherwise be highly susceptible to predation if immediately killed or immobilised (Hawkins, 1994). As a consequence, koinobionts tend to have shorter lifespans (Gauld and Bolton, 1988) and are usually specialists with narrower host ranges than idiobionts (Askew and Shaw, 1986; Hawkins, 1994), and so tend to dominate external herbivore communities (Hawkins, 1994). In the current

study, neither the abundance nor the taxon richness of koinobiont taxa was apparently influenced by the field margin treatments. This suggests that any increased botanical diversity associated with the margin treatments (Sheridan *et al.*, 2008) had not yet reached a point where it started to substantially influence the diversity of externally feeding arthropod hosts for koinobiont parasitoids. Indeed with the possible exception of aphids and perhaps other sap-sucking Hemiptera, grassland may not be a habitat conducive to the incidence of a biodiverse, externally feeding phytophagous insect fauna. In contrast, however, the observed increase in the diversity and abundance of idiobiont taxa suggests that the greatest advantage of a field margin in an agricultural grassland context, at least in the short term, is associated with a rapid increase in plant structural diversity (elongated stems, flowers, seed heads, etc.) not normally evident in intensively grazed and cut agricultural grasslands, which provides greatly enhanced opportunities for well-concealed, cryptic plant-mining insect populations that are a suitable resource for substantially increased idiobiont populations.

Good bioindicators can be used not only to reflect overall biodiversity but also to measure and document ecological change within the environment in which they are found. They should reflect the relevant change by their changing presence and/or relative abundance, and have a high probability of being sampled when present in a given circumstance (McGeoch, 1998). Indicator value analysis showed that the parasitoid genus *Polynema* (egg parasitoids of cicadellid leaf-hoppers (Huber, 1997)) is a good potential indicator genus for intensively grazed agricultural grassland, whilst *Inostemma*, a genus of parasitoids attacking cecidomyiid gall-midge eggs and larvae, and *Panstenon*, a predator of plant-hoppers (Delphacidae), were good potential indicators of the structurally more complex grassland habitat provided by the fenced field margins. (Clearly supporting evidence for the bioindicator value of these parasitoid genera will be reported by the wider Ag-Biota Project, which found that cicadellid leaf-hoppers of the genus *Macrostelus* were significantly more abundant in the grazed paddock margins, whilst the delphacid genus, *Javesella*, was more abundant in fenced field margins.) In contrast, and as reported above, very few

Lepidoptera could be found in vegetation suction samples taken from this field margin experiment, and yet the guild of lepidopterous parasitoids was found to be one of the best biological indicators for ROT margins compared with normally grazed paddock margins. This example illustrates probably better than any other the unique potential and bioindicator value of parasitoid taxa for cryptic, 'hidden' biodiversity that is not easily documented by any other means. Although there were relatively few genera with high (>70%) IVs, three genera (*Polynema*, *Inostemma* and *Panstenon*) had very strong associations with their habitat (IVs greater than 85%). It is not unusual for only a few taxa to be indicative of a particular habitat, but if parasitoid identification had been possible to the species level, more indicator taxa might have been observed.

This study has shown that conserving field margins in intensively managed grassland (REPS 3 Measure 2)

leads to increased diversity and abundance of parasitoid Hymenoptera. Idiobionts and parasitoids of cryptic plant-mining hosts particularly benefit from such a measure. Since many parasitoid taxa are important natural enemies of agricultural crop pests, and constitute an extremely valuable plant protection resource within agro-ecosystems, this finding has potentially important functional, as well as conservational, significance. Ongoing analysis of the data provided by our monitoring of the conservation field margin experiment will seek to better understand the parasitoid–host relationships of the taxa recorded in the field margins, and to use such insights to better explain the ecological processes and consequences of a conservation strategy that even the most intensive grassland farmers in Ireland might readily undertake at minimal cost to their production system.

5 The Impacts of Farm Management Practices on the Diversity and Ecology of Parasitoid Hymenoptera

5.1 Background

The management of agricultural grasslands throughout Europe has become more intensive through increased fertiliser inputs and stocking rates, and this has led to landscape degradation and loss of biodiversity (McLaughlin and Mineau, 1995; Duelli, 1997; Buchs, 2003; Hoffman and Greef, 2003). It is important to know how these changing grassland management practices impact on biological diversity. Increased nitrogen application leads to decreased plant species richness (Tilman, 1987; Plantureux *et al.*, 2005), which in turn can lead to decreased insect herbivore species richness (Haddad *et al.*, 2000; Plantureux *et al.*, 2005). Heavy grazing also reduces sward structure, leading to limited shelter and opportunities for foraging activity for invertebrates (Purvis and Curry, 1981; Morris, 2000). Reduced production intensity, including the reduction of nutrient inputs and consequent stocking rates, can produce favourable conditions for increased faunal diversity through increased plant botanical and structural diversity for herbivorous populations and an increased accumulation of vegetation for decomposer communities (Plantureux *et al.*, 2005).

One of the main aims of agri-environmental schemes is to increase biodiversity. The CAP has made provisions for agri-environmental schemes amongst its member states and these schemes aim to provide farmers with incentives to carry out environmentally friendly changes (Ovenden *et al.*, 1998). The REPS, initiated on 1 June 1994, was Ireland's response to this concept (Emerson and Gillmor, 1999) and is designed to reward farmers for environmental improvements made to their farms, and for their management of the wider environment. Measure 2 in the current third incarnation of REPS aims to promote a sustainable grassland management plan protecting habitats, minimising poaching, overgrazing and soil erosion.

Measure 1 provides for a nutrient management plan, which specifies a total overall limit on nutrient inputs to agricultural grassland of 260 kg N/ha, including both organic and inorganic sources (Department of Agriculture and Food, 2006).

In this chapter we use two existing grassland husbandry experiments at Teagasc Research Centres to investigate more closely the use of parasitoid diversity and community structure as bioindicators of resulting ecological effects on wider biodiversity. The first of these experiments at Johnstown Castle was designed to investigate the agronomic effects of reducing the level of nitrogen fertiliser application, and the second was set up by Teagasc at their research centre to compare the agronomic performance of a conventional system and a less-intensive REPS-compatible suckler beef production system.

5.2 Aims

The aims of our investigation of these experiments were to:

- Investigate the response of parasitoids to the cessation of nitrogen inputs (extensification) in a paddock-based rotational grazing system experiment at Teagasc, Johnstown Castle (Tower Field experiment)
- Investigate the response of parasitoids in a paddock-based comparison between a conventional, high-input system and a REPS-based beef husbandry system (Grange experiment)
- Integrate the results from these studies into a preliminary assessment of the influence of grassland management practice on parasitoid biodiversity in an agricultural grassland context.

5.3 Methods

5.3.1 *The response of parasitoids to nitrogen inputs – Tower Field experiment*

5.3.1.1 *Experimental design*

The biodiversity and community structure of parasitoids were investigated within a field plot experiment at Johnstown Castle, Co. Wexford, Ireland (T015173), to study the effects of reduced nitrogen use. The experiment was established in 2001 on an imperfectly drained gley loam soil over clay loam derived from Irish Sea till (Gardiner and Ryan, 1964). Prior to the commencement of the experiment, the field in question contained a uniform perennial ryegrass sward that had been established for more than 10 years, and had been intensively managed for commercial beef production with fertiliser application of c. 350 kg N/ha/year. The experiment, arranged in a randomised block design, comprised three replicate paddocks of three treatments with N fertiliser rates of 0, 225 and 390 kg/ha/year, respectively. The nitrogen fertiliser was applied as urea (46% N) for the first two applications in spring and as calcium ammonium nitrate (CAN – 26% N) applied in remaining applications from May to September. Paddock size varied with treatment (from 0.4 ha for the 390 kg N treatment to 1.0 ha for the 0 kg N treatment) in order to accommodate a common 21-day rotational grazing cycle (7 days grazing, 14 days recovery) by a self-contained herd of 10–15 steers within each treatment. This equated to stocking rates of 1, 2.4 and 3.0 livestock units, respectively, for the 0, 225 and 390 kg N treatments. Paddocks were cyclically strip-grazed with each main paddock being divided into three strips with back fencing for this purpose. Unfortunately, after the experiment had been established for only 1 year, one of the three 0-N paddocks was found to be unrepresentatively waterlogged, and so was dropped from the experiment.

5.3.1.2 *Parasitoid collection*

Parasitoids were collected from the experimental paddocks using a Vortis Insect Suction Sampler (Burkard Manufacturing Co. Ltd, Rickmansworth, Hertfordshire, UK) (Arnold, 1994) in 2004, 4 years after the start of the experiment. A single pooled sample was taken from four randomly selected sampling spots

within each grazing strip, each spot sampled for 10 s. This provided three nested samples from each (main paddock) treatment replicate. Sampling was carried out in three consecutive weeks during May (12th, 18th and 25th) and July/August (28th, 4th and 9th, respectively). On each sampling occasion, mean sward height within each paddock strip was assessed using a Jenquip Plate Meter. The total area sampled with the Vortis sampler in each paddock on each sampling occasion was 0.075 m², and the total area sampled over the entire programme of sampling was 0.45 m² per paddock. Catches were preserved in 70% ethanol prior to sorting and the identification of parasitoids to the level of genus using the taxonomic literature listed in Table 2.1.

5.3.1.3 *Statistical analysis*

All analyses were carried out using R version 2.4.1 (R Development Core Team, 2006). Treatment comparisons of the taxon richness and total abundance of parasitoids were made using glmmPQL with a Poisson error structure in view of the count nature of the data. Because of the system of nested sampling that was adopted within a randomised block experiment, treatment, block and grass height were modelled as fixed effects, whilst sampling date and individual grazing strip within main paddocks were treated as random effects. The maximal model containing all factors was fitted first, and then simplified by sequential removal of non-significant terms. The minimal adequate model was determined using AIC values (Crawley, 2005).

5.3.1.4 *Community structure and ecology of parasitoids*

The host range of parasitic Hymenoptera collected from the Tower Field was identified using published literature. Parasitoids were grouped into guilds as follows: idiobionts, koinobionts, parasitoids of Coleoptera, Diptera, Hemiptera (Aphididae), Hemiptera (other than aphids), Lepidoptera, fungal-feeding insect larvae, dung-breeding insect larvae, gall-forming insect larvae, and eggs of spiders and insects. The effects of the experimental treatments on parasitoid community structure was investigated using NMDS implemented by the PC-ORD program (McCune and Mefford, 2006) (see Section 4.3.3.3 for further details). The data for the individual grazing strips within the main paddocks were pooled across

sampling dates and transformed ($\log_{10}(x+1)$) prior to analysis. NMDS using Sorensen Distance was run in the autopilot mode with a random starting configuration and 50 runs with the data. A Monte Carlo test was used to evaluate whether the extracted axes differed from random chance, and to determine the most appropriate dimensionality (McCune and Mefford, 2006).

5.3.1.5 Indicator values

Indicator values were calculated for parasitoid genera and guilds collected from each of the nitrogen treatments. Indicator values were calculated by pooling data collected from each main treatment paddock across all sampling dates. See Section 4.3.3.4 for further details concerning the method of IV calculation.

5.3.2 Comparison of parasitoids in REPS-compatible and conventional beef systems – the Grange experiment

5.3.2.1 Experimental design

The experiment was originally established at the Grange Teagasc Farm in 2002 with the aim of comparing agronomic aspects of a conventional intensive spring-calving suckler-beef production system with a similar REPS-compatible system. The two systems were operated with their own individual suckler herds and differed with respect to a wide range of animal husbandry aspects, including fertiliser use, stocking rate and silage conservation (Table 5.1).

The field experiment was set up on two large blocks of land, each containing two randomly allocated fields for each treatment. Thus the experiment contained four replicates of each treatment. Each replicate field was divided into three nested grazing paddocks, which

were rotationally grazed in a synchronised sequence, grazing one field of each treatment at any one time.

5.3.2.2 Parasitoid collection

Parasitoids and other sward arthropods were sampled using a Vortis Insect Suction Sampler (Burkard Manufacturing Co. Ltd, Rickmansworth, Hertfordshire, UK) (Arnold, 1994) in August 2003, 18 months after the start of the experiment. One aggregate sample was taken from each grazing paddock within main treatment fields, these samples consisting of ten randomly selected subsamples, each of 10 s duration. This provided three nested samples from each of the eight main treatment fields. At the time of sampling, mean sward height was assessed within each grazing strip using a Jenquip Plate Meter. Catches were preserved in 70% ethanol prior to sorting and the identification of parasitoids to the level of genus using the taxonomic literature listed in Table 2.1.

5.3.2.3 Data analysis

Treatment comparisons of parasitoid taxon richness and abundance were made using glmmPQL, using the R statistical programme, version 2.4.1 (R Development Core Team, 2006). A Poisson error structure was specified because of the count nature of the data. Because of the system of nested sampling within a randomised block experiment, treatment, block and grass height were modelled as fixed effects and individual grazing strip was treated as a random effect. A maximal model containing all factors was fitted first, and then simplified by sequential removal of non-significant terms. The minimal adequate model was determined using AIC values (Crawley, 2005).

5.3.2.4 Community structure and ecology of parasitoids

The host range of parasitic Hymenoptera collected from the Grange experiment was identified using

Table 5.1. Stocking rates and animal husbandry in the conventional and REPS-compatible systems compared in the Grange experiment.

	Conventional	REPS
Total inorganic nitrogen per season	225 kg N/ha	88 kg N/ha
Stocking rate	0.65 ha/cow unit ¹	0.82 ha/cow unit
Silage cuts per season	2	1
Total harvested silage area	0.69 ha/cow unit	0.56 ha/cow unit
Percentage grassland harvested for forage per season	85%	55%

¹1 cow unit = 1 suckler cow + progeny to slaughter + 25% replacements per season.

published literature. Parasitoid taxa were grouped into the following guilds: idiobionts, koinobionts, parasitoids of aphids, Hemiptera (other than aphids), Lepidoptera, Coleoptera, Diptera, fungal-feeders, dung-breeders, gall-formers, and egg parasitoids. The effects of the beef system treatments on parasitoid community structure were investigated using NMDS using the PC-ORD program (McCune and Mefford, 2006) (see Section 4.3.3.3 for further details). The catches from all samples collected in each grazing paddock were pooled and the data were transformed to $\log_{10}(x+1)$ before NMDS was run using the Sorenson Distance measure in the autopilot mode with a random starting configuration, and 50 runs with the data. A Monte Carlo test was used to assess whether the extracted axes differed from random chance, and to help select the best dimensionality (McCune and Mefford, 2006).

5.3.2.5 Indicator values

Indicator values were calculated for the parasitoid genera and guilds collected from each beef system, pooling all catches made in each grazing paddock. See Section 4.3.3.4 above for further details concerning the calculation of IVs.

5.4 Results

5.4.1 Tower Field experiment

5.4.1.1 Abundance and taxon richness of parasitoid taxa in the Tower Field experiment

A total of 1,820 individuals from 72 genera were collected from a total sampled area of 5.95 m² in the Tower Field experiment. The results of the generalised linear mixed model fitted to parasitoid abundance data are summarised in Table 5.2. There were more parasitoid individuals in samples from the 0 N treatment than from either the 225 N or 390 N

treatments (Table 5.2; Fig. 5.1). There was no difference in parasitoid abundance between 225 N and 390 N treatments.

Two parasitoid taxa in particular, *Polynema* (Mymaridae) and *Trimorus* (Scelionidae), were observed to be very abundant in samples from the Tower Field experiment, and both these taxa were significantly more abundant in the 0 N treatment (Table 5.3, Figs 5.2 and 5.3). Both these genera are parasitoids of insect eggs, specifically the eggs of cicadellid leaf-hoppers and mirid bugs in the case of *Polynema* (Huber, 1997), and more widely of the eggs on insects and spiders in the case of *Trimorus* (Polaszek and Notton, 2003). The genus *Macrosteles* Fieber (Cicadellidae) was one of the most abundant insect genera encountered in samples from the Tower Field experiment and analysis of the abundance of this likely host group for both *Polynema* and *Trimorus* found the same pattern of abundance with significantly higher numbers of *Macrosteles* found in the 0 N compared with the 225 N or 390 N treatments (Table 5.3; Fig. 5.4).

In contrast to clearly significant treatment effects on parasitoid abundance (numbers of individuals) in samples from the nitrogen input treatments in the Tower Field experiment, no significant treatment difference was found in the taxon richness (numbers of genera) of parasitoids collected (Fig. 5.5).

5.4.1.2 Community structure and ecology of parasitoids in the Tower Field experiment

A complete list of genera collected from the Tower Field experiment with details of their host ranges obtained from the literature is given in Table 5.4. The majority of these taxa are parasitoids of plant-mining

Table 5.2. Summary of results¹ from analysis of the effects of nitrogen input treatments on the abundance of parasitoid individuals in the Tower Field experiment at Teagasc, Johnstown Castle, Co. Wexford. There were no significant interactions and only the significant effects obtained from the minimal model are shown.

Parameter	Parameter estimate	Standard error	df	t	p
0 N	2.0674	0.4223	135	4.895	<0.0001
225 N	-0.6156	0.1056	135	5.832	<0.0001
390 N	-0.7443	0.1085	135	6.858	<0.0001

¹An initial maximal model included nitrogen treatment, block and grass height as fixed effects, and sampling date and grazing strip as random effects. Only details for the final minimal model are shown.

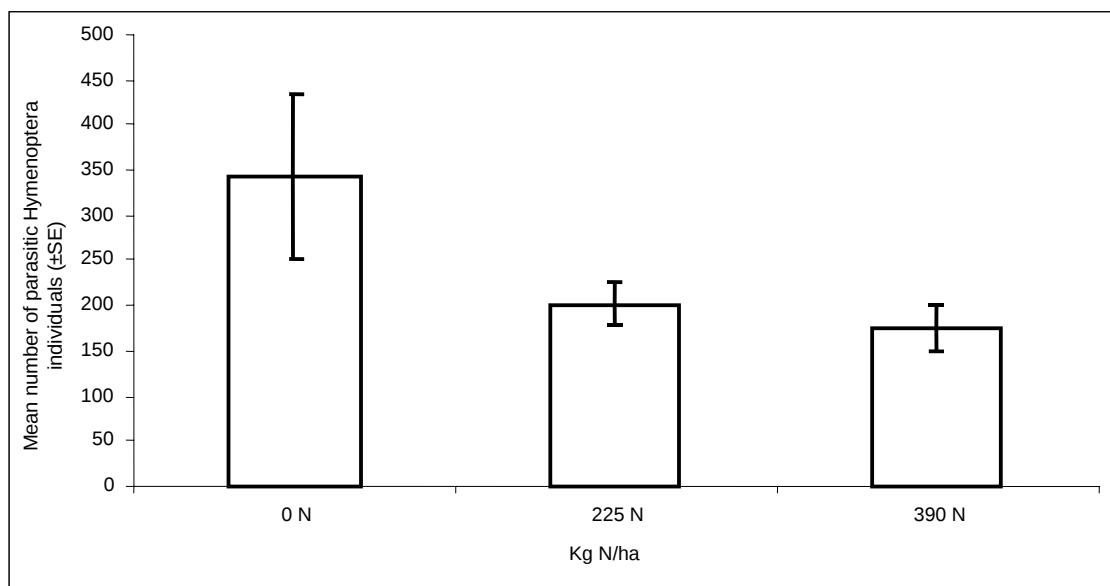


Figure 5.1. Estimated mean numbers of parasitoid individuals in Vortis suction samples from a sampled area of 0.45 m² collected from the nitrogen input treatments in the Tower Field experiment, Johnstown Castle.

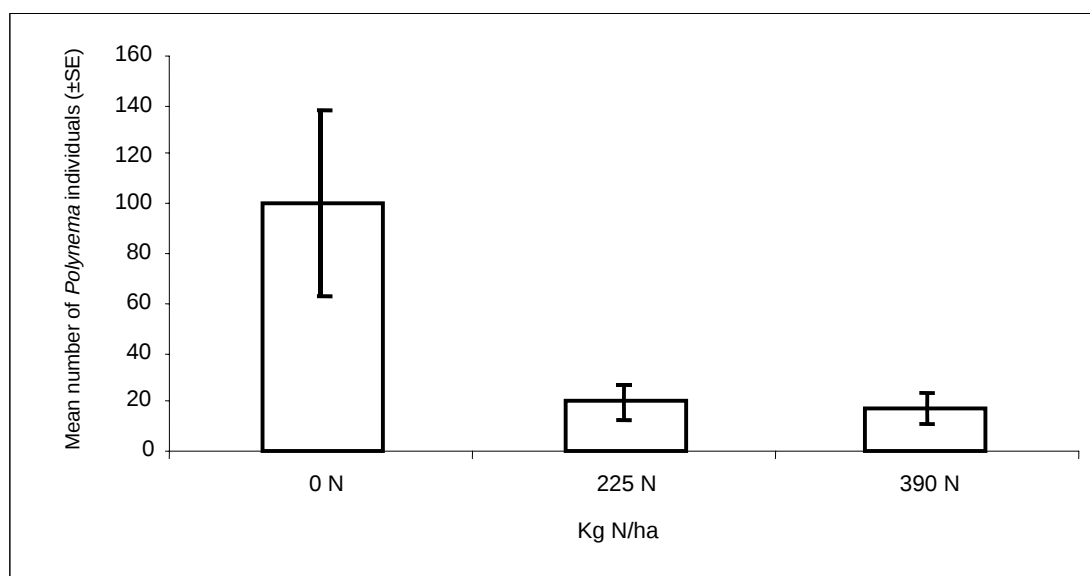


Figure 5.2. Estimated mean numbers of the hemipteran egg parasitoid *Polynema* (Mymaridae) collected in Vortis suction samples from the nitrogen input treatments in the Tower Field experiment in 2004.

and/or gall-forming insects, especially Diptera, although other prominent host groups include aphids and other sap-sucking Hemiptera, dung- and fungal-feeding Diptera, Lepidoptera, some Coleoptera, and eggs of various arthropod groups including insects and spiders. Parasitoids of holometabolous groups

(Coleoptera, Diptera and Lepidoptera) are generally parasitoids of immature larval, pupal or egg life stages.

NMDS analysis of community structure using individual parasitoid genera, produced a final three-dimensional ordination with a stress of 13.42 and a

Table 5.3. Summary results¹ from analyses of the effects of nitrogen input treatments on the abundance of the parasitoid genera *Polynema* and *Trimorus* and one of their most abundant potential hosts *Macrosteles* (Hemiptera: Cicadellidae).

Response variable	Final t statistics for treatment comparisons (df)		
	0 vs 225 N	0 vs 390 N	225 vs 390 N
<i>Polynema</i>	−7.836 (134)***	−8.135 (134)***	NS
<i>Trimorus</i>	−4.274 (134)***	−5.695 (134)***	NS
<i>Macrosteles</i> nymphs	−9.515 (135)***	−10.588(135)***	NS

¹A maximal model was initially fitted with treatment, block and grass height included as fixed effects and grazing paddock and sampling date as random effects. For each response variable, a minimal adequate model was derived by stepwise removal of non-significant terms. In each case, a final *t* value was calculated for pairwise treatment comparisons using the minimum adequate model: NS, non-significant; ****p* < 0.001.

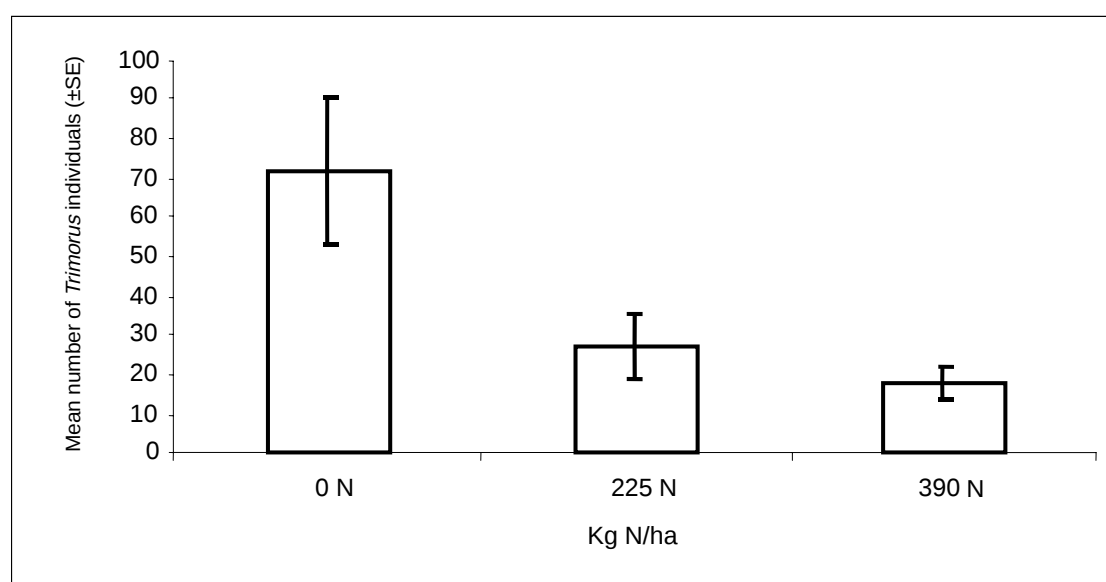


Figure 5.3. Estimated mean numbers of the insect and spider egg parasitoid genus *Trimorus* (Scelionidae) collected in Vortis suction samples from the nitrogen input treatments in the Tower Field experiment in 2004.

final instability of 0.00001 from 109 iterations. A final stress of between 10 and 20 can correspond to a meaningful ordination, but values towards the upper end of this range need careful interpretation (Clarke, 1993). A Monte Carlo test based on 50 randomised runs gave *p* values of 0.0196 for each of the first three axes, indicating that the ordination was unlikely to have been obtained by chance. Axes 1 and 2 accounted for 36.2% and 34.9% of the variance (approximately 71% in combination), respectively, and show a clear separation of parasitoid populations in paddocks of the 0 N treatment compared with those of the 225 N and 390 N treatments (Fig. 5.6). The genera *Gonatocerus*

(likely parasites of cicadellid leaf-hopper eggs) and *Meraporus* (parasitoids of plant-mining weevil and gall-midge larvae) were more closely associated with paddocks of the 0 N treatment. All other genera, including *Aphidius*, *Monoctonus* and *Trioxys* (parasitoids of aphids), *Dendrocercus* and *Alloxysta* (hyperparasitoids of aphids; primary parasitoids of Hymenoptera such as *Aphidius*, *Monoctonus* and *Trioxys*), *Dinotrema*, *Phaenocarpa* and *Pentapleura* (parasitoids of dung or fungal-feeding Diptera larvae), and *Chorebus*, *Chasmodon* and *Platygaster* (parasitoids of plant-mining or gall-forming Diptera larvae or eggs), appear to be more associated with the

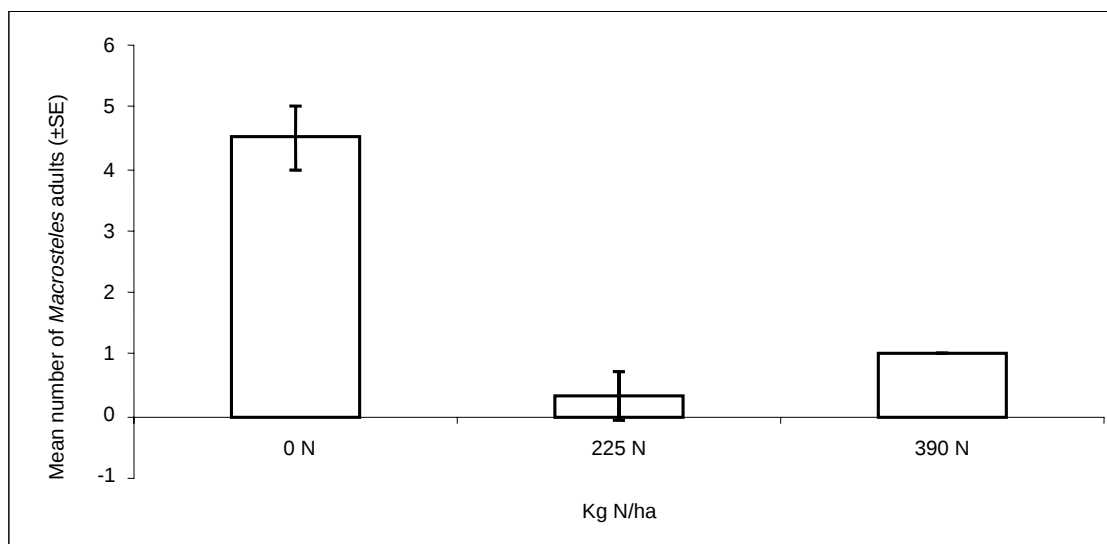


Figure 5.4. Estimated mean numbers of the cicadellid leaf-hopper genus *Macrosteles* (likely hosts for *Polynema* and *Trimorus* egg parasitoids) collected in Vortis suction samples from the nitrogen input treatments in the Tower Field experiment in 2004.

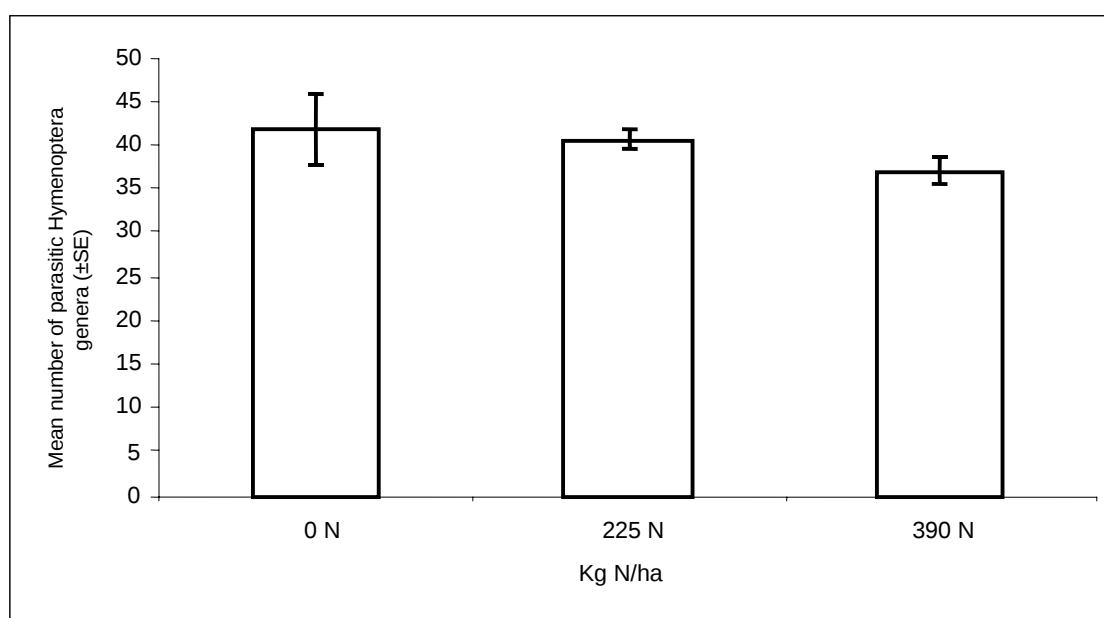


Figure 5.5. Estimated mean numbers of parasitoid Hymenoptera genera collected in Vortis suction samples from the nitrogen input treatments of the Tower Field experiment, Johnstown Castle.

paddocks receiving nitrogen fertiliser applications (Fig. 5.6).

NMDS ordination of parasitoid guilds produced a three-dimensional solution with a final stress of 9.448 and a final instability of 0.00001 with 69 iterations. A final stress of between 5 and 10 gives a good

ordination with no real risk of drawing false inferences (Clarke, 1993). A Monte Carlo test gave a p-value of 0.0196 for the first three axes, indicating that the ordination was unlikely to have been obtained by chance. Axes 1 and 2 accounted for 23.9% and 44.8% of the variance in the data (approximately 68% in combination), respectively, and show a separation of

Table 5.4. Summary of information concerning the known host ranges of parasitoid taxa collected from the Tower Field experiment.

Family	Genus	Host range	Reference
Aphelinidae	<i>Aphelinus</i>	Aphididae (Hemiptera)	Woolley, 1997
Braconidae	<i>Aphidius</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Ephedrus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Monoctonus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Trioxys</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Coelinidea</i>	Plant-mining Chloropidae Diptera larvae	Wharton, 1997a
Braconidae	<i>Blacus</i>	Coleoptera larvae	Sharkey, 1997
Braconidae	<i>Opius</i>	Diptera larvae	Wharton, 1997c
Braconidae	<i>Pentapleura</i>	Dung-breeding Diptera	Wharton, 1997a
Braconidae	<i>Phaenocarpa</i>	Dung or fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Aspilota</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Dinotrema</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Orthostigma</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Chasmodon</i>	Leaf-mining Diptera larvae	Umoru, 1997
Braconidae	<i>Microplitis</i>	Lepidoptera larvae	Whitfield, 1997
Braconidae	<i>Chorebus</i>	Plant-mining Diptera larvae	Wharton, 1997a
Braconidae	<i>Dacnusa</i>	Plant-mining Diptera larvae	Wharton, 1997a
Braconidae	<i>Dapsilarthra</i>	Plant-mining Diptera larvae	Wharton, 1997a
Ceraphronidae	<i>Aphanogmus</i>	Gall-forming Cecidomyiidae larvae (Diptera)	Alekseev, 1987
Ceraphronidae	<i>Ceraphron</i>	Gall-forming Cecidomyiidae larvae (Diptera)	Alekseev, 1987
Diapriidae	<i>Trichopria</i>	Diptera larvae	Nixon, 1980
Diapriidae	<i>Basalys</i>	Fungal-feeding Diptera larvae	Nixon, 1980
Diapriidae	<i>Pamis</i>	Fungal-feeding Diptera larvae	Nixon, 1957
Dryinidae	<i>Gonatopus</i>	Homoptera (Hemiptera) nymphs	Ponomarenko, 1987
Encyrtidae	<i>Copidosma</i>	Lepidoptera larvae	Noyes <i>et al.</i> , 1997
Encyrtidae	<i>Rhopus</i>	Pseudococcidae (Hemiptera)	Noyes <i>et al.</i> , 1997
Eulophidae	<i>Aprostocetus</i>	Gall-forming Cecidomyiidae larvae (Diptera)	Schauff <i>et al.</i> , 1997
Eulophidae	<i>Hemiptarsenus</i>	Leaf-mining insect larvae	Askew, 1968b
Eulophidae	<i>Diglyphus</i>	Plant-mining Diptera larvae	Askew, 1968b
Eulophidae	<i>Closterocerus</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera, Homoptera)	Schauff <i>et al.</i> , 1997
Eulophidae	<i>Pediobius</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera)	Schauff <i>et al.</i> , 1997
Eurytomidae	<i>Tetramesa</i>	Gall-forming insect larvae	Di Giulio, 1997
Figitidae	<i>Melanips</i>	Diptera larvae	Fergusson, 1986
Figitidae	<i>Kleidotoma</i>	Dung and fungal-feeding Diptera	Quinlan, 1978
Figitidae	<i>Phaenoglyphis</i>	Hyperparasitoid via Aphidiinae	Fergusson, 1986
Figitidae	<i>Alloxysta</i>	Hyperparasitoids via Aphidiinae	Fergusson, 1986
Figitidae	<i>Rhoptromeris</i>	Leaf-mining insect larvae	Quinlan, 1978
Ichneumonidae	<i>Phygadeuon</i>	Diptera pupae	Townes, 1970a
Ichneumonidae	<i>Campoletis</i>	Lepidoptera larvae	Azidah <i>et al.</i> , 2000
Ichneumonidae	<i>Stenomacrus</i>	Fungal-feeding Mycetophilidae (Diptera) larvae	Townes, 1971
Ichneumonidae	<i>Aclastus</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera)	Townes, 1970a

Table 5.4 contd.

Family	Genus	Host range	Reference
Ichneumonidae	<i>Gelis</i>	Various cocoons (e.g. Lepidoptera, Hymenoptera, spiders)	Townes, 1970a
Megaspilidae	<i>Dendrocerus</i>	Hyperparasites via Aphidoidea	Fergusson, 1980
Megaspilidae	<i>Conostigmus</i>	Syrphidae	Alekseev, 1987
Mymaridae	<i>Ooctonus</i>	Cercopidae, Cicadellidae (Hemiptera) eggs	Huber, 1997
Mymaridae	<i>Gonatocerus</i>	Cicadellidae and Membracidae (Hemiptera) eggs	Huber, 1997
Mymaridae	<i>Polynema</i>	Cicadellidae, Miridae (Hemiptera) eggs	Huber, 1997
Mymaridae	<i>Anagrus</i>	Hemiptera eggs	Huber, 1997
Mymaridae	<i>Anaphes</i>	Various insect eggs	Huber, 1997
Platygastridae	<i>Inostemma</i>	Cecidomyiidae eggs or larvae	Kozlov, 1987
Platygastridae	<i>Platygaster</i>	Cecidomyiidae eggs or larvae	Kozlov, 1987
Platygastridae	<i>Synopeas</i>	Cecidomyiidae eggs or larvae	Kozlov, 1987
Proctotrupidae	<i>Codrus</i>	Coleoptera larvae in soil and litter	Kozlov, 1987
Pteromalidae	<i>Semiotellus</i>	Cecidomyiidae pupae	Bouček and Rasplus, 1991
Pteromalidae	<i>Meraporus</i>	Curculionidae (Coleoptera), Cecidomyiidae (Diptera) larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Spalangia</i>	Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Asaphes</i>	Hyperparasites via Aphidiinae	Bouček and Rasplus, 1991
Pteromalidae	<i>Mesopolobus</i>	Plant-miners	Bouček and Rasplus, 1991
Pteromalidae	<i>Seladerma</i>	Plant-miners	Bouček and Rasplus, 1991
Pteromalidae	<i>Callitula</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Cyrtogaster</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Halticoptera</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Panstenon</i>	Predators of Delphacidae (Hemiptera) eggs	Bouček and Rasplus, 1991
Pteromalidae	<i>Trichomalopsis</i>	Various pupae (e.g. Lepidoptera, Coleoptera, Diptera, Hymenoptera)	Bouček and Rasplus, 1991
Scelionidae	<i>Telenomus</i>	Insect and spider eggs	Polaszek and Notton, 2003
Scelionidae	<i>Trimorus</i>	Insect and spider eggs	Polaszek and Notton, 2003

parasitoid guild structure in paddocks of the 0 N treatment compared with those of the 225 N and 390 N treatments (Fig. 5.7). The parasitoids of Hemiptera (other than aphids) and insect and spider eggs tended to be more associated with the 0 N treatment. Conversely, parasitoids of dung-breeding insects, Diptera, aphids and Coleoptera were more strongly associated with paddocks receiving nitrogen fertiliser applications (Fig. 5.7).

5.4.1.3 Indicator values for parasitoid taxa in the Tower Field experiment

When comparing the 0 N treatment with the combined 225 N and 390 N treatments (henceforth referred to as higher N), the genera *Polynema* (parasitoids of cicadellid eggs), *Gonatocerus* (parasitoids of leaf-

hopper eggs), *Trimorus* (parasitoids of insect and spider eggs), *Meraporus* and *Aprostocetus* (both parasitoids of gall-forming and plant-mining dipteran larvae) were found to be a good potential indicator of the 0 N treatments with a significant ($p < 0.05$) IV greater than 70% (Table 5.5). The genus *Platygaster* (parasitoids of insect eggs) had significant ($p < 0.05$) associations with the 0 N paddocks, but their IV of 67.8 was somewhat lower (Table 5.5). When contrasting populations in the 0 N and higher N paddocks, no parasitoid genus had an IV for the latter treatment greater than 70%; however, the genera *Phaenocarpa* (parasitoids of dung- or fungal-feeding dipterous larvae), *Chorebus* and *Aphanogmus* (parasitoids of plant-mining Diptera), and *Aphidius* and *Trioxys* (aphid parasitoids) were all significantly associated with

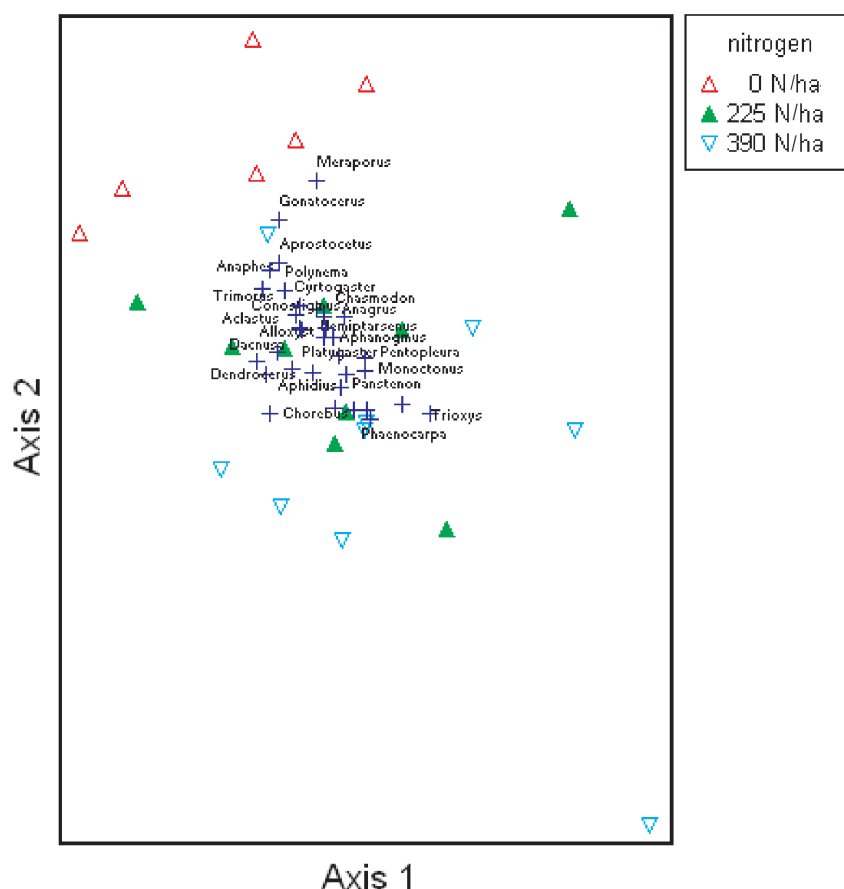


Figure 5.6. NMDS ordination of parasitoid Hymenoptera genera collected from paddocks of the Tower Field experiment at Johnstown Castle in 2004. Axes 1 and 2 account for 36.2% and 34.9% of the variance, respectively. Crosses represent the ordination of individual parasitoid genera relative to that of the treatment paddocks.

higher N paddocks with IVs in the range 55–68% (Table 5.5).

Indicator value analysis for parasitoid guilds collected from the Tower Field experimental paddocks showed that several guilds were significantly associated with the 0 N paddocks in comparison with the higher N treatments, including parasitoids of plant gall-forming larvae, various insect larvae and eggs and Hemiptera other than aphids. Parasitoids of aphids were significantly associated with the higher N treatments in comparison with the 0 N paddocks (Table 5.6).

5.4.2 Grange experiment

5.4.2.1 Abundance and genera richness of parasitoid taxa collected from the Grange experiment

A total of 726 individuals representing 42 genera of parasitoid Hymenoptera were collected from a total

sampled area of 1.44 m² in the Grange experiment. There was no significant treatment difference in the mean abundance of parasitoids (Fig. 5.8), nor was there a difference in the mean taxon richness of parasitoids collected from the conventional and REP-compatible systems (Fig. 5.9).

5.4.2.2 Community structure, host range and biology of parasitoids collected from the Grange experiment

A complete list of genera collected from the Grange experiment with details of their host ranges obtained from the literature is given in Table 5.7. The majority of these taxa are parasitoids of aphids and plant-mining insects, although other hosts include dung- and fungal-feeding dipteran larvae, coleopteran larvae and pupae, Hemiptera (other than aphids) and the eggs of insects and spiders.

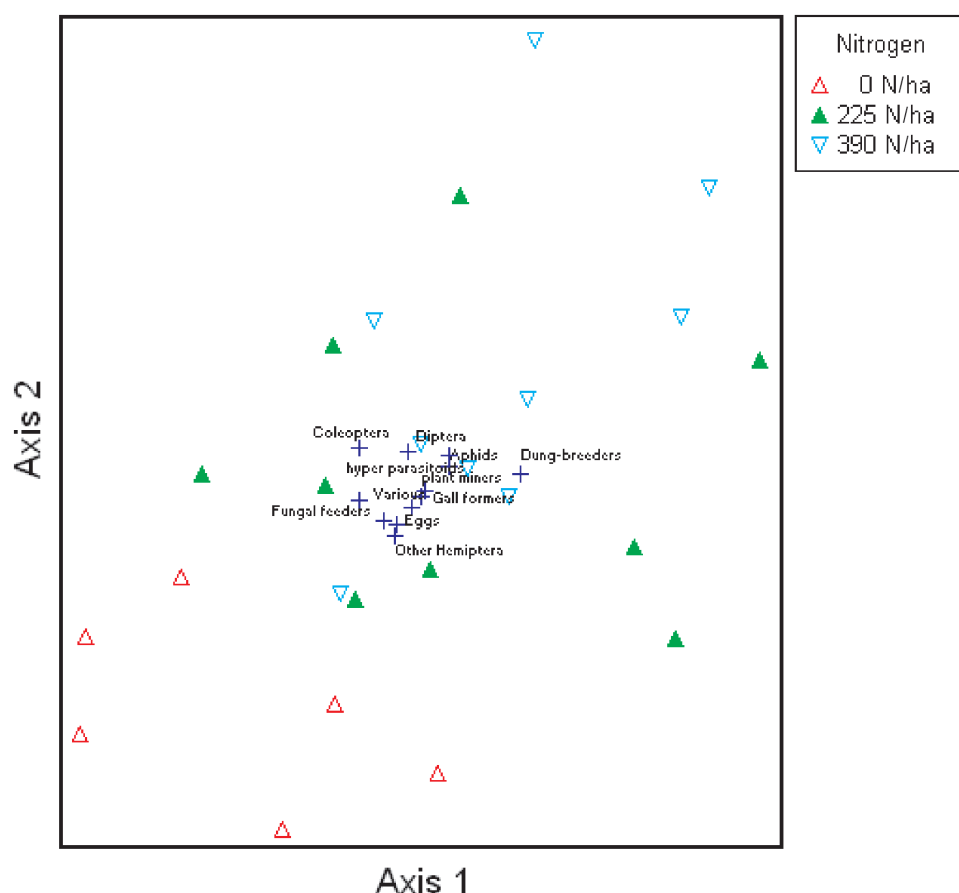


Figure 5.7. NMDS ordination for parasitoid wasp guilds collected from the Tower Field experiment at Johnstown Castle in 2004. Axes 1 and 2 account for 23.9% and 44.8% of data variance, respectively. Crosses represent the guild groupings relative to that of treatment paddocks.

Table 5.5. Comparative indicator values¹ for parasitoid genera collected from the nitrogen input treatments in the Tower Field experiment in 2004. 0 N treatments were compared to the combined 225 N and 390 N (higher N) treatments.

Genus	Parasitoids of	Indicative of	IV	p-value
<i>Aphanogmus</i>	Gall-forming Cecidomyiidae larvae	Higher N	63.2	0.0084
<i>Aphidius</i>	Aphididae (Hemiptera)	Higher N	66.7	0.0630
<i>Aprostocetus</i>	Gall-forming Cecidomyiidae larvae	0 N	75.7	0.0074
<i>Chorebus</i>	Plant-mining Diptera larvae	Higher N	65.7	0.0300
<i>Gonatocerus</i>	Cicadellidae (Hemiptera) eggs	0 N	75.0	0.0036
<i>Meraporus</i>	Plant-mining Curculionidae and gall-forming Cecidomyiidae larvae	0 N	72.1	0.0028
<i>Phaenocarpa</i>	Dung- or fungal-feeding Diptera larvae	Higher N	65.5	0.0378
<i>Platygaster</i>	Gall-forming insect eggs or larvae	0 N	67.8	0.0184
<i>Polynema</i>	Cicadellidae (Hemiptera) eggs	0 N	84.5	0.0012
<i>Trimorus</i>	Insect and spider eggs	0 N	76.2	0.0028
<i>Trioxys</i>	Aphididae (Hemiptera)	Higher N	55.6	0.0478

¹Indicator values were calculated for pooled catches from individual paddocks. Only genera with significant ($p = 0.05$) IVs are shown; taxa with significant IVs $\geq 70\%$ (emboldened) can be considered representative of the treatment they are associated with (McGeoch *et al.*, 2002).

Table 5.6. Comparative indicator values¹ for parasitoid guilds collected from the nitrogen input treatments in the Tower Field experiment in 2004. 0 N treatments were compared to the combined 225 N and 390 N treatments.

Parasitoid guild	Associated with	IV	p-value
Parasitoids of:			
Plant gall-forming insect larvae	0 N	54.0	0.0056
Hemiptera (other than aphids)	0 N	59.8	0.0006
Various insect larvae	0 N	58.9	0.0022
insect and spider eggs	0 N	57.9	0.0004
Hemiptera: Aphididae	Higher N	62.3	0.0042

¹Indicator values were calculated for pooled catches from individual paddocks. Only guilds with significant ($p = 0.05$) IVs are shown (McGeoch *et al.*, 2002).

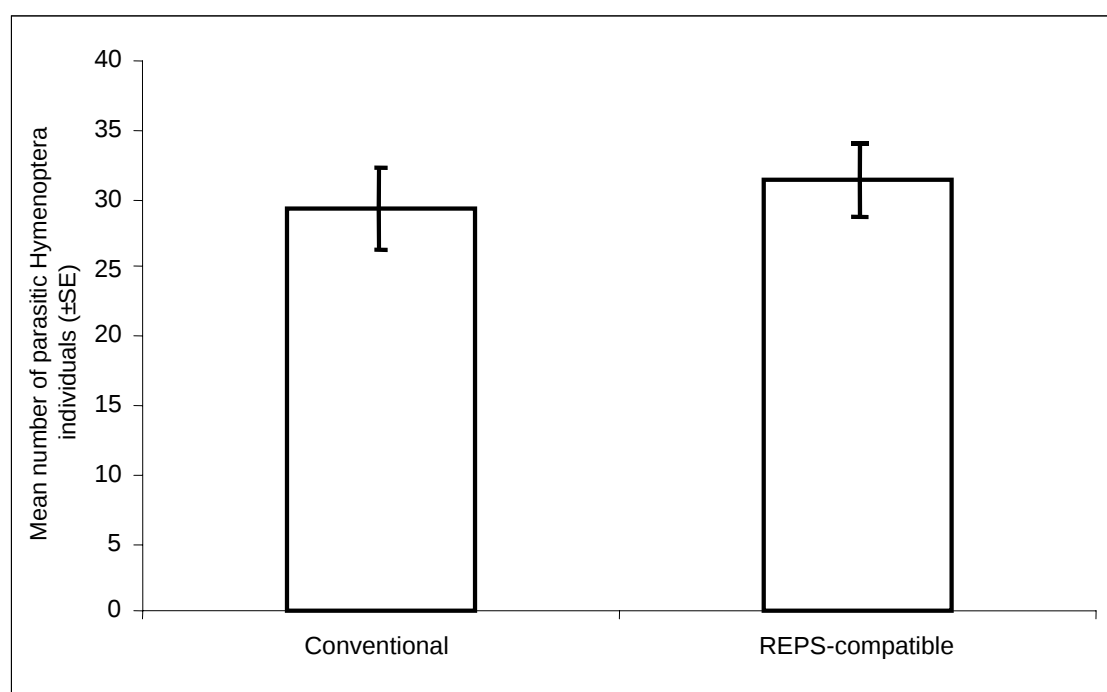


Figure 5.8. Estimated mean numbers of parasitoid individuals collected in Vortis suction samples from the conventional and REPS-compatible beef production systems in the Grange experiment.

An NMDS analysis of community structure in the Grange experiment using individual parasitoid genus data produced a final three-dimensional ordination with a stress of 15.28 and a final instability of 0.00001 from 102 iterations. A final stress of between 10 and 20 corresponds to a meaningful ordination, but values towards the upper end of this range need careful interpretation (Clarke, 1993). A Monte Carlo test on 50 randomised runs gave a p-value of 0.0196 for the first two axes and 0.0392 for the third axis, indicating that the final ordination was unlikely to have been obtained by chance. Axes 1 and 2 accounted for 21.7% and

47.2% of the variance (approximately 70% in combination), respectively, and this plot shows a tendency to separate conventional and REPS-compatible fields on Axis 2; however, the groups of conventional and REPS-compatible samples do overlap (Fig. 5.10). The genera *Polynema* (likely parasites of cicadellid leaf-hopper eggs), *Meraporus* (parasites of plant-mining weevil and cecidomyiid gall-midge larvae), and *Pentapleura* (parasites of dung-feeding dipteran larvae) tended to be associated with REPS-compatible fields. Conversely, the genera *Diglyphus* (parasites of plant-mining Diptera), *Aphidius*

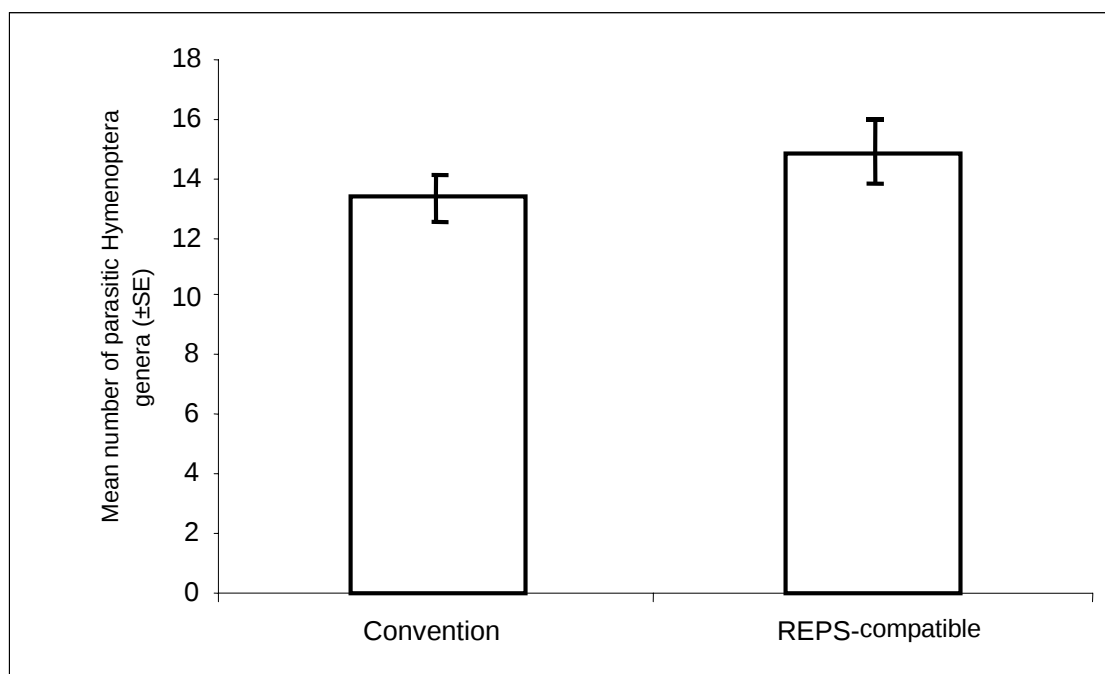


Figure 5.9. Estimated mean numbers of parasitoid genera collected in Vortis suction samples from the conventional and REPS-compatible beef production systems in the Grange experiment.

and *Trioxys* (both parasites of aphids) tended to be associated with conventional fields (Fig. 5.10).

The analysis of parasitoid community structure in the conventional and REPS-compatible fields using parasitoid guild data (based on host ranges) produced a final three-dimensional ordination with a stress of 10.67 with a final instability of 0.00001 from 92 iterations. A Monte Carlo test gave p-values of 0.0196 for the first two axes and 0.0392 for the third axis, indicating that the final stress was unlikely to have been obtained by chance. Axes 1 and 2 accounted for 45.3% and 26.2% of the variance in the data (approximately 71% in combination), respectively, and show single strongly outlying samples from each system at opposite ends of Axis 1, but in general show a large degree of overlap between system samples (Fig. 5.11).

5.4.2.3 Indicator values for parasitoid taxa and guilds in the Grange beef system experiment

In comparing parasitoid populations in the two experimental beef systems, only two genera had significant IVs (Table 5.8); *Diglyphus* was significantly more associated with the conventional system, whilst *Meraporus* was more associated with the REPS-

compatible system. The IVs for these taxa, however, were only 59.1% and 52.1%, respectively, so neither can be considered a good indicator of systems they were associated with.

5.5 Discussion

The cessation of nitrogen inputs to agricultural grassland in the Tower Field experiment clearly benefited both the abundance and taxon richness of parasitoid Hymenoptera and, by inference from the demonstrated relationship between parasitoid incidence and the diversity of other arthropod taxa, such a reduction in nitrogen use is likely also to have benefits for a much wider range of arthropods. Two parasitoid genera in particular, *Polynema* and *Trimorus*, were strongly associated with the 0 N treatment. *Polynema* is a parasitoid of the eggs of cicadellid leaf-hoppers, whilst *Trimorus* is a genus of more generalist egg parasitoids of insects and spiders. Both are potentially parasitoids of the eggs of *Macrosteles* spp., the immature nymphs of which were found to be one of the most abundant insect populations in the 0 N paddocks. Parasitoids of dung-breeding Diptera were clearly associated with paddocks of the higher nitrogen treatments. This is

Table 5.7. Parasitoid genera collected from Grange beef system experiment and their host ranges.

Family	Genus	Host range	Reference
Aphelinidae	<i>Aphelinus</i>	Aphididae (Hemiptera)	Woolley, 1997
Braconidae	<i>Aphidius</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Ephedrus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Monoctonus</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Praon</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Trioxys</i>	Aphididae (Hemiptera)	Achterberg, 1997
Braconidae	<i>Coelinidea</i>	Plant stem-mining Chloropidae larvae (Diptera)	Wharton, 1997a
Braconidae	<i>Blacus</i>	Coleoptera larvae	Sharkey, 1997
Braconidae	<i>Opius</i>	Diptera larvae	Wharton, 1997c
Braconidae	<i>Pentapleura</i>	Dung-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Phaenocarpa</i>	Dung- or fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Aspilota</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Dinotrema</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Orthostigma</i>	Fungal-feeding Diptera larvae	Wharton, 1997a
Braconidae	<i>Chasmodon</i>	Leaf-mining Diptera larvae	Umoru, 1997
Braconidae	<i>Chorebus</i>	Plant-mining Diptera larvae	Wharton, 1997a
Braconidae	<i>Dacnusa</i>	Plant-mining Diptera larvae	Wharton, 1997a
Braconidae	<i>Dapsilarthra</i>	Plant-mining Diptera larvae	Wharton, 1997a
Ceraphronidae	<i>Aphanogmus</i>	Gall-forming Cecidomyiidae larvae (Diptera)	Alekseev, 1987
Diapriidae	<i>Trichopria</i>	Diptera larvae	Nixon, 1980
Encyrtidae	<i>Lamennaisia</i>	Hemiptera	Noyes <i>et al.</i> , 1997
Eulophidae	<i>Aprostocetus</i>	Gall-forming Cecidomyiidae larvae (Diptera)	Schauff <i>et al.</i> , 1997
Eulophidae	<i>Hemiptarsenus</i>	Leaf-mining insect larvae	Askew, 1968b
Eulophidae	<i>Diglyphus</i>	Plant-mining Diptera larvae	Askew, 1968b
Eulophidae	<i>Chrysocharis</i>	Various (e.g. Lepidoptera, Diptera, Hymenoptera)	Schauff <i>et al.</i> , 1997
Figitidae	<i>Phaenoglyphis</i>	Hyperparasitoid via Aphidiinae	Fergusson, 1986
Figitidae	<i>Alloxysta</i>	Hyperparasitoids via Aphidiinae	Fergusson, 1986
Figitidae	<i>Rhoptromeris</i>	Leaf-mining insect larvae	Quinlan, 1978
Ichneumonidae	<i>Phygadeuon</i>	Diptera pupae	Townes, 1970a
Ichneumonidae	<i>Stenomacrus</i>	Mycetophilidae (fungal-feeding Diptera larvae)	Townes, 1971
Ichneumonidae	<i>Aclastus</i>	Various (e.g. Lepidoptera, Coleoptera, Hymenoptera)	Townes, 1970a
Ichneumonidae	<i>Gelis</i>	Various cocoons (e.g. Lepidoptera, Hymenoptera, spiders)	Townes, 1970a
Megaspilidae	<i>Dendrocerus</i>	Hyperparasites via Aphidoidea	Fergusson, 1980
Megaspilidae	<i>Conostigmus</i>	Syrphidae	Alekseev, 1987
Mymaridae	<i>Gonatocerus</i>	Cicadellidae and Membracidae eggs (Hemiptera)	Huber, 1997
Mymaridae	<i>Polynema</i>	Cicadellidae and Miridae eggs (Hemiptera)	Huber, 1997
Mymaridae	<i>Anagrus</i>	Hemiptera eggs	Huber, 1997
Mymaridae	<i>Anaphes</i>	Various eggs	Huber, 1997
Platygastridae	<i>Platygaster</i>	Gall-forming Cecidomyiidae eggs or larvae	Kozlov, 1987
Platygastridae	<i>Leptacis</i>	Gall-forming Cecidomyiidae eggs or larvae	Kozlov, 1987
Proctotrupidae	<i>Codrus</i>	Coleoptera larvae in soil litter	Kozlov, 1987
Pteromalidae	<i>Meraporus</i>	Plant-mining Curculionidae (Coleoptera) and gall-forming Cecidomyiidae (Diptera) larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Spalangia</i>	Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Mesopolobus</i>	Plant-mining insect larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Seladerma</i>	Plant-mining insect larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Cyrtogaster</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Stenomalina</i>	Plant-mining Diptera larvae	Bouček and Rasplus, 1991
Pteromalidae	<i>Trichomalopsis</i>	Various pupae (e.g. Lepidoptera, Coleoptera, Diptera, Hymenoptera)	Bouček and Rasplus, 1991
Scelionidae	<i>Telenomus</i>	Insect and spider eggs	Polaszek and Notton, 2003
Scelionidae	<i>Trimorus</i>	Insect and spider eggs	Polaszek and Notton, 2003

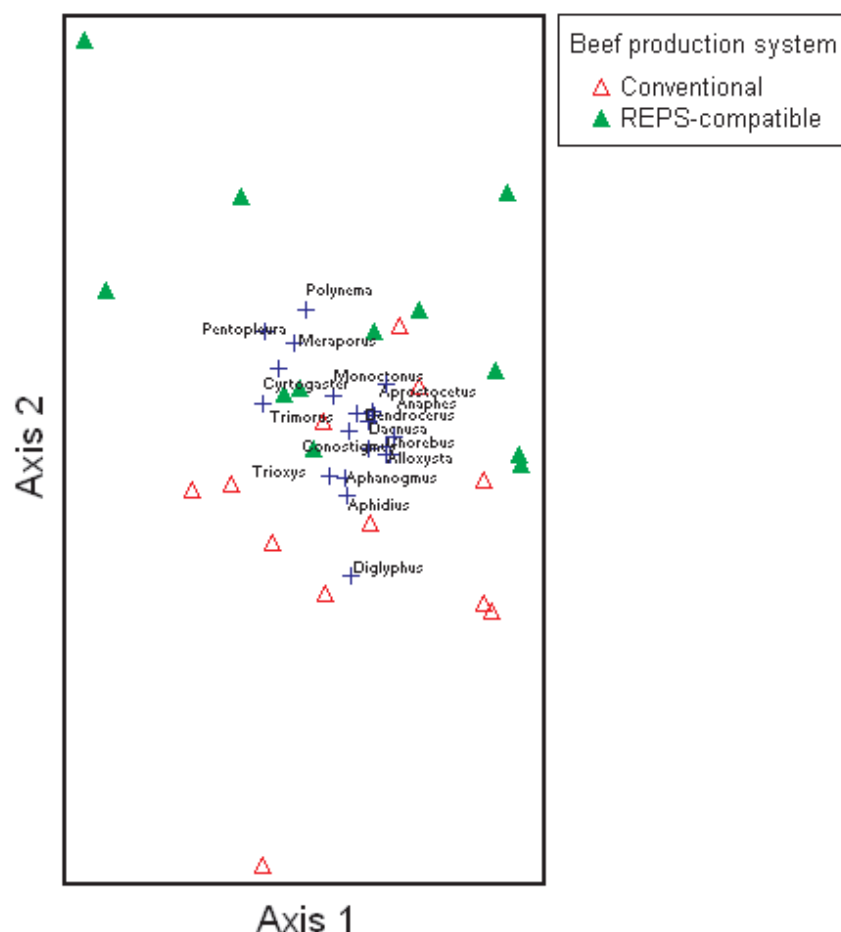


Figure 5.10. NMDS ordination of parasitoid Hymenoptera genera collected from rotational grazing paddocks of the Grange beef system experiment in 2003. Axes 1 and 2 account for 21.7% and 47.2% of the variance, respectively. Crosses represent the ordination of individual parasitoid genera relative to that of system fields.

Table 5.8. Comparative indicator values¹ (IVs) of parasitoid genera/guilds collected from the Grange beef experiment in 2003.

Genus	Parasitoids of	Associated with	IV	p-value
<i>Diglyphus</i>	Plant-mining Diptera larvae	Conventional system	59.1	0.0064
<i>Meraporus</i>	Plant-mining Curculionidae and gall-forming Cecidomyiidae larvae	REPS-compatible	52.1	0.0498

¹Indicator values were calculated for pooled catches from each field. Only taxa with significant ($p = 0.05$) IVs are shown; no group had a sufficiently large IV ($\geq 70\%$) to be considered representative of either system (McGeoch *et al.*, 2002).

entirely consistent with the increased incidence of cattle dung that might be expected on the more heavily stocked paddocks of the 225 N and 390 N treatments.

Analysis of community structure on the paddocks of the Tower Field experiment using NMDS ordination showed that the cessation of nitrogen inputs had more subtle effects on both the taxonomic composition and

the guild structure of parasitoid populations. These changes suggest that they were due to changes in relative incidence of parasitoid host populations. Thus, both the genera and guild structure of parasitoids associated with dung-breeding insect larvae, Diptera (including many dung-breeders) and sap-sucking aphids were more prominent in the high-input nitrogen

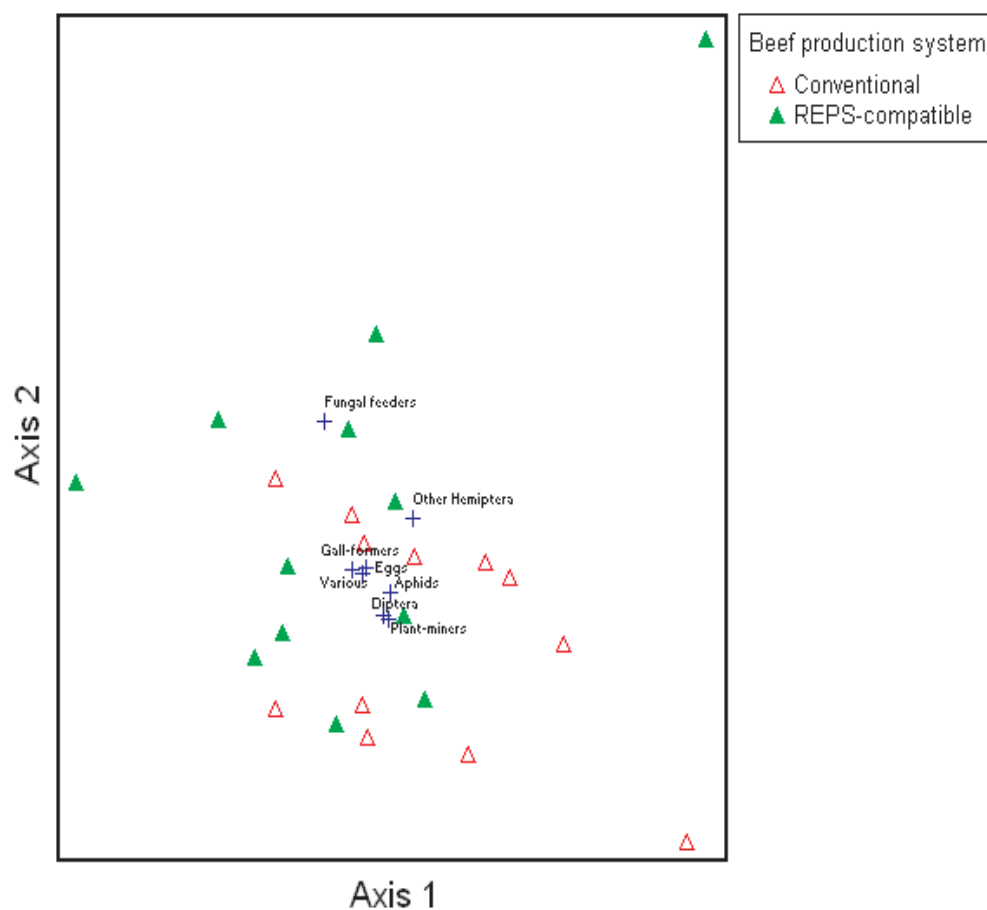


Figure 5.11. NMDS ordination of parasitoid guilds collected from rotational grazing paddocks of the Grange beef system experiment in 2003. Axes 1 and 2 account for 45.3% and 26.2% of the variance, respectively. Blue crosses represent the ordination of guilds relative to that of system fields.

treatments, whereas parasitoid populations in the 0 N paddocks had a greater prominence of parasitoid populations associated with non-aphid hemipteran bugs, fungal-feeding insect larvae and insect and spider eggs. This shift in parasitoid community structure indicates an ecological broadening of overall arthropod community structure, beyond that dominated by dung-feeders and specialised sap-suckers in intensive, high nutrient input systems. Indicator value analysis confirmed this widening of ecological roles in the 0 N treatment, significantly identifying several parasitoid genera of plant-mining and gall-forming insect larvae, and insect and spider eggs, as good indicators of the 0 N input treatment and a genus of dung-breeding Diptera (*Phaenocarpa*) as characteristic of very high nitrogen inputs.

Responses by parasitoid Hymenoptera to the experimental contrast of conventional and REPS-

compatible beef production systems were less obvious. However, NMDS ordination of parasitoid community structure using information on genera composition revealed a similar pattern of diverging parasitoid community structure in the less-intensive REPS-compatible system, with again a shift towards greater prominence of parasitoids associated with concealed plant-mining and galling insect hosts and egg parasitoids in the less-intensive system, and aphid parasitoids (*Aphidius* and *Trioxys*) more evident in the intensive system. Effects in the Grange experiment were smaller and much subtler. This may well reflect the relatively smaller absolute difference in treatment contrasts in the two experiments (0 vs 390 kg N at Tower Field, and 88 vs 225 kg N at Grange). However, both experiments were essentially comparisons of reduced nutrient inputs. Neither experiment departed from the widely practised system of intensive rotational grazing in fenced paddock strips, which maximises

grass utilisation. Undoubtedly, this is likely to have limited the potential beneficial effects of the less-intensive treatments on both sward botanical and arthropod biodiversity. Insect diversity and taxon richness decline when botanical structural diversity is reduced (Lawton, 1983), and intensive grazing greatly

reduces the numbers of grassland insects (Morris, 1967). In contrast, taller grassland supports a much greater abundance and diversity of arthropods as it permits the development of a much wider range of trophic niches (Morris, 2000).

6 General Discussion

6.1 Increasing the Taxonomic Resolution of Studies on Parasitoid Hymenoptera

A total of 178 genera of parasitoid Hymenoptera were identified in these studies of agricultural grasslands and verified by independent taxonomic experts. These included ten taxa not previously recorded from Ireland. All taxa, including the new records, were collected from moderately-to-intensively managed grasslands, suggesting that even in the most commonplace of habitats, there may be much Irish biodiversity as yet to be discovered. Perhaps even more importantly, the project has resulted in the development of the necessary local expertise, and the development of essential international contacts with taxonomic specialists, to make it possible to study further this very important group of arthropods.

6.2 Parasitoid Hymenoptera as Bio-indicators of Wider Arthropod Diversity

Analysis of the relationship between parasitoid wasp incidence and the diversity of all other arthropod taxa using data from three independent data sets (the initial Ag-Biota study of just ten paired grassland sites, the much larger Ag-Biota study of 50 grassland sites and the study of the Teagasc conservation field margin experiment at Johnstown Castle) all showed that the incidence of parasitoid wasps was significantly, and better related to the diversity of all other arthropod taxa than was the incidence of any other individual arthropod group. Whilst a degree of caution is always advisable when interpreting correlations from single data sets, the consistency of the relationships between parasitoid wasp incidence and the diversity of other arthropods in the studied grasslands provides convincing support for the original hypothesis that, because of their unique biologies, parasitoid wasps are good indicators of the diversity of all other arthropod taxa. Despite the improving level of taxonomic knowledge and literature availability to facilitate the identification of parasitoid wasps, parasitoid

identification will always require a quite specialised taxonomic skill, and so would be logistically difficult to integrate into any realistic framework of widespread monitoring. It is therefore particularly relevant that, with the exception of the much smaller data set provided by the initial Ag-Biota study of just ten sites, the total abundance of parasitoid individuals was actually found to be better related to overall arthropod taxon richness than was the number of parasitoid genera.

The routine sampling and quantification of total parasitoid abundance (total numbers of individuals) would be a relatively much more straightforward and practicable option for routine monitoring. There is therefore a very reasonable prospect that parasitoid wasps could be used as bioindicators to track ongoing changes in arthropod diversity, at least within the context of agricultural grasslands, which comprise the greater proportion of the Irish countryside. Whilst monitoring changes in the abundance of parasitoid populations might be used to document loss or improvement in wider arthropod biodiversity, it would contribute relatively little to explaining the likely causes of such change. However, our studies have shown that knowledge of the detailed composition and community structure of parasitoid populations, particularly with respect to the relative incidence of idiobiont and koinobiont developmental biologies and guilds with different host-group affinities, can provide a much more detailed insight into the underlying ecological processes causing change in wider arthropod community structure. To gain this insight, however, requires a much more detailed understanding of the biologies and relative abundance of individual parasitoid taxa.

6.3 The Indicator Value of Parasitoid Community Structure in Agricultural Grasslands

The Teagasc field margin experiment clearly showed that fencing the margins of intensively managed dairy paddocks, preventing grazing and the application of all inputs to the sward, creates conditions in which

substantially more parasitoid genera and individuals can be found compared with the unfenced margins of normal grazed paddock. This effect is very likely to be a reflection of the greater plant structural diversity, and perhaps also altered microclimatic conditions in the ungrazed margins (Purvis and Curry, 1981). When the relative abundance of different parasitoid taxa were compared in the fenced and normal paddock margins, taxa that parasitise very well concealed and hidden hosts such as Hemiptera (other than aphids), cecidomyiid gall-midge larvae, plant-mining insect larvae and parasitoids of the eggs of insects and spiders, were found to benefit most from fencing. One of the most distinctive indicator groups for the field margins was the guild of lepidopteran larval parasitoids. As virtually no adult Lepidoptera were collected from any of the experimental margins during the study, it is very likely that the majority of lepidopterous larval parasitoids observed in the conservation margins were parasites of well concealed, plant-mining, 'micro-lepidopteran' larvae. Observation of their parasitoids is probably the only feasible way that the presence of such cryptic biodiversity within grassland plants can be measured, and our data infer a much greater prevalence of such hosts in fenced margins. This example illustrates, probably better than any other, the unique potential of parasitoid taxa as bioindicators of cryptic, 'hidden' biodiversity that is not easily documented by any other means.

The idiobiont/koinobiont dichotomy also provided important evidence of a greater prevalence of concealed insect hosts in the fenced margin treatments. The idiobiont developmental strategy seems to be particularly well adapted to the parasitism of hosts that are not likely to be taken by other predators following parasitism (Hawkins, 1994). In the current study, both the abundance and taxon richness of idiobionts were substantially greater in the structurally more diverse fenced margin treatments compared to the unfenced paddock margins, again supporting the conclusion that the main advantage of the fenced treatments was to create more niche opportunities for concealed hosts that mine within the greater variety of plant structures that are available in ungrazed conditions. In contrast, it is likely that koinobiont parasitoids have become much more

closely adapted to their host's development in order to keep the latter alive and avoid predation, which would otherwise be highly likely in an exposed host that was killed or immobilised immediately following parasitism (Hawkins, 1994). In the current study, neither the abundance nor the taxon richness of koinobiont taxa was apparently influenced by the field margin treatments, suggesting that any increased botanical diversity associated with the fenced margin treatments had not yet reached a point where it had started to influence the abundance or diversity of exposed hosts suitable for koinobiont parasitoids. With the possible exception of aphids and other sap-sucking Hemiptera, agricultural grassland may not be a habitat immediately conducive to the incidence of a biodiverse, externally feeding phytophagous insect fauna. In contrast, however, the marked increase in the diversity and abundance of idiobiont taxa observed in fenced margins suggests that the main advantage of conservation field margins (or extensive grazing systems) in an agricultural grassland context, at least in the short term, is the rapid and obvious increase in plant structural diversity (elongated stems, flowers, seed heads, etc.). This enhanced plant structural diversity appears to provide increased opportunities for well-concealed, cryptic plant-mining/galling larval insect populations that constitute suitable hosts for idiobiont parasitoids, that are much less prevalent in intensively grazed and cut swards.

6.4 Parasitoid Assemblages as Indicators of the Intensity of Management Practice

The cessation of nitrogen inputs to agricultural grassland in the Tower Field experiment clearly benefited both the abundance and taxon richness of parasitoid Hymenoptera and, by inference from the demonstrated relationship between parasitoid incidence and the diversity of other arthropod taxa, such an extreme reduction in nitrogen use is likely also to enhance the abundance and diversity of a wider range of arthropods. In particular, the genera *Polynema* (parasitoids of the eggs of cicadellid leafhoppers) and *Trimorus* (more generalist egg parasitoids of insects and spiders) were significantly more abundant in paddocks receiving the reduced, 0 N treatment. Indicator value analysis showed that both

these genera and a range of other parasitoid taxa utilising concealed plant-mining and galling insect hosts and non-aphid Hemiptera were significantly associated with the 0 N treatment. In contrast, the genera *Phaenocarpa* (parasitoids of dung-breeding Diptera) and *Aphidius* and *Trioxys* (parasitoids of aphids) were significantly associated with paddocks receiving the higher nitrogen treatments. This is entirely consistent with the likely increased incidence of cattle dung, and a higher incidence of aphids (Dixon, 1997) that might be expected on the more heavily stocked paddocks that received high nitrogen inputs.

Analysis of community structure on the paddocks of the Tower Field experiment showed that the cessation of nitrogen inputs also had more subtle effects on both the taxonomic composition and the guild structure of parasitoid populations. These effects again suggested that the fundamental basis of effects on parasitoid assemblages were predictable differences in the relative incidence of potential host populations. Thus, both the genera and guild structure of parasitoids associated with dung-breeding insect larvae and sap-sucking aphids were more prominent in the high-input nitrogen treatments, whereas the 0 N paddocks had a greater prominence of parasitoid populations associated with non-aphid hemipteran bugs, fungal-feeding insect larvae and insect and spider eggs. This shift in parasitoid assemblages clearly indicates an ecological broadening of overall arthropod community structure, beyond that dominated by dung-feeders and specialised sap-suckers typical of intensive, high nutrient input systems.

The response of parasitoid Hymenoptera populations to the experimental contrast of conventional and REPS-compatible beef production systems at Teagasc, Grange was less obvious. However, NMDS ordination of parasitoid community structure using information on taxon (genera) composition, revealed the same basic tendency towards diverging parasitoid community structure in the less-intensive REPS-compatible system, with again a similar underlying shift towards a greater predominance of parasitoid taxa associated with concealed plant-mining and galling insect hosts and egg parasitoids in the latter system, and a greater prominence of aphid parasitoids (*Aphidius* and *Trioxys*) in the more intensive system. In

comparison to effects seen in the Tower Field experiment, however, the effects of the Grange REPS-compatible beef system were relatively small and much more subtle.

6.5 Implications for Current REPS Policy

Taken together, the evidence from all three Teagasc field experiments (the paddock margin experiment, Tower Field nitrogen input experiment and the Grange beef system experiment), tell a rather similar story of effects on parasitoid abundance, diversity and altered parasitoid community structure, parasitoid taxa associated with predictable host groups, such as aphids and dung-breeding Diptera, being more prominent in the most intensive grassland treatments, while a greater diversity of parasitoids of particularly well-concealed plant-mining and gall-forming hosts and parasitoids of insect and spider eggs being characteristically more prominent in extensive treatments. However, the degree to which such treatment differences were apparent was effectively a reflection of the relative contrast in the 'intensity' of management systems being compared. Thus, by far the clearest discrimination of parasitoid populations was observed in the comparison of experimental field margin treatments, in which populations in all the fenced treatments differed very clearly from those in the UC margins (differences between fenced treatments themselves being small). Within the Tower Field experiment, differences in parasitoid populations were greatest between the most (390 kg N/ha/year) and the least (0 kg N/ha/year) intensive nutrient input treatments, but even these were relatively small in comparison to the differences seen in the conservation margin experiment. Treatment effects in the Grange beef system experiment were even smaller.

Collectively, these results are very revealing and clearly support previous work showing the importance of sward structure on arthropod populations in relatively intensively managed agricultural grasslands (Purvis and Curry, 1981). This dimension of grassland management was only evident in the field margin experiment. The large plot experiments at Tower Field (Johnstown Castle) and Grange essentially provided only comparisons of reduced nutrient inputs and

consequently reduced stocking intensities, as neither experiment departed from the widely practised system of intensive rotational grazing of fenced paddock strips, which maximises grass utilisation. Undoubtedly, this is likely to have limited the potential biodiversity benefits of the less-intensive grassland management in terms of reduced grazing intensity. These studies highlight a possible deficiency in current REPS policy with respect to grassland management. As the Grange beef system experiment clearly illustrates, it is possible for farmers to manage their grassland according to REPS specifications by reducing only nitrogen use and stocking rates. However, if the pattern of grass utilisation by intensive rotational grazing and cutting for forage remains unchanged, the net effect of such measures on biodiversity is likely to be small. However, simply fencing field margins, and thus preventing grazing and chemical inputs, was shown to increase arthropod biodiversity in these intensively managed landscapes. More detailed studies of alternative grazing and grass utilisation systems are required to optimise the potential biodiversity benefits of REPS policy with respect to grassland husbandry.

6.6 Final Conclusions

Our studies originally set out to test the hypothesis that the parasitoid Hymenoptera could provide a useful means of assessing wider arthropod diversity in agro-ecosystems. There is a wide range of theoretical arguments to support such a claim, including the high trophic position of these taxa within the wider arthropod communities in which they occur, and the

unique nature of their biological relationships with practically all other major insect groups. Our studies quite clearly illustrate this bioindicator potential, and we believe confirm the initial hypothesis. Using multiple data sets, we have shown that both the abundance and diversity of parasitoid wasp taxa correlate with overall arthropod diversity in agricultural grasslands better than any other group. While the fact that their simple abundance is a reliable indicator of overall arthropod diversity provides us with a practicable and eminently usable monitoring tool, we have also shown that using a more detailed knowledge of their taxonomy, biology and host groups, we can gain a unique insight into underlying influences affecting wider arthropod community structure. This insight more than justifies the necessary taxonomic effort required to identify parasitoids at the level of genus. However, because of taxonomic difficulties, ecologists have traditionally regarded the structure of parasitoid wasp communities as almost impossible to study. However, this project has shown that taxonomic knowledge is no longer a serious limitation, and the use of parasitoid Hymenoptera as ecologically sensitive indicators of the environmental effects of specific farm husbandry practices is now an achievable reality. With improving knowledge of their host group relationships, parasitoids offer a unique means to study highly 'cryptic' hidden biodiversity, which would otherwise be almost impossible to assess directly by any other means, even within the most commonplace of habitats.

7 References

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Science, Technology, Research and Innovation for the Environment (STRIVE) 2007-2013

The Science, Technology, Research and Innovation for the Environment (STRIVE) programme covers the period 2007 to 2013.

The programme comprises three key measures: Sustainable Development, Cleaner Production and Environmental Technologies, and A Healthy Environment; together with two supporting measures: EPA Environmental Research Centre (ERC) and Capacity & Capability Building. The seven principal thematic areas for the programme are Climate Change; Waste, Resource Management and Chemicals; Water Quality and the Aquatic Environment; Air Quality, Atmospheric Deposition and Noise; Impacts on Biodiversity; Soils and Land-use; and Socio-economic Considerations. In addition, other emerging issues will be addressed as the need arises.

The funding for the programme (approximately €100 million) comes from the Environmental Research Sub-Programme of the National Development Plan (NDP), the Inter-Departmental Committee for the Strategy for Science, Technology and Innovation (IDC-SSTI); and EPA core funding and co-funding by economic sectors.

The EPA has a statutory role to co-ordinate environmental research in Ireland and is organising and administering the STRIVE programme on behalf of the Department of the Environment, Heritage and Local Government.